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Front Cover: *Utricularia minor*, Eldorado County, California, USA. Photo by Barry Rice.
Article on page 194.

Back Cover: *Cephalotus* flowers at different stages from blooming to releasing seeds.
Image by John Brittnacher. Article on page 168.

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A NEW SCHEDULE OF FEES FOR ICPS STARTING FEBRUARY 2023

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Printing and mailing costs of CPN have continued to increase, and despite our best efforts to keep the prices unchanged, the ICPS must pass these increased costs on to members to ensure we remain financially viable. CPN and other member services still provide excellent value for carnivorous plant enthusiasts, and our education and conservation programs are a valuable way of increasing awareness of carnivorous plants and protecting vulnerable species in their natural habitats. We hope that members will continue to support these endeavours despite the modest fee increases:

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OBSERVATIONS ON *CEPHALOTUS FOLLICULARIS* REPRODUCTIVE BIOLOGY

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Keywords: *Cephalotus follicularis*, flowers, seeds, seed germination, stolons.

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Abstract: Aspects of *Cephalotus follicularis* reproductive biology are documented to gain a better understanding of this enigmatic plant. Detailed observations of seed germination, seedling development, and vegetative reproduction provide a description of the uncommon events in the process.

Cephalotus follicularis (Cephalotaceae, Oxalidales) is found in bog and swamp margins, fens, and seeps in southwestern Australia. The area has a winter rainy season and summer dry season. Localized flooding can occur during the winter. Bushfires are an integral part of the ecology of southwestern Australia during the dry season. *Cephalotus* has particular adaptations to survive overgrowth by shrubs and to take advantage of the fires that remove the overgrowth (DeBuhr 1976; Lowrie 1978; Gibson 1999; Mann 2005; Nunn 2014; Cross *et al.* 2019). The plants ecological adaptations are reflected in its reproductive biology.

Materials and methods

The *Cephalotus* plants used in this study were initially grown from seeds donated to the ICPS Seed Bank in 2000 and 2006 by Phill Mann or were the cultivars *Cephalotus* ‘OG Black’ and *Cephalotus* ‘Hummer’s Giant’. The plants had been rejuvenated multiple times from stolon cuttings. They were grown under natural light by a garage window supplemented with artificial lighting on a local sunrise/sunset cycle to induce flowering. Flowers were pollinated by hand and by flies. Observations of the flowering process were made to make seed production more productive. Seven flowers were followed closely. Achenes were collected and seeds removed to determine seed set.

The seedling study was done either planting seeds in pots with finely chopped long fibered New Zealand *Sphagnum* or in plastic petri dishes with filtration disks. For the petri dishes, attempts were made to adequately sterilize the seeds and filtration disks using sulfur powder and a hydrogen peroxide solution. What worked best was dusting sulfur powder onto filtration disks, putting another disk over and two under it to give a stack of four disks to hold moisture. The stacks were placed in the petri dishes with the same inside dimensions as the diameter of the disks. Seeds were placed in a 1% hydrogen peroxide solution in a microtiter tube for 5 minutes then the seeds and solution poured onto the disks in the petri dishes. Additional water was added as necessary. The dishes were put in plastic Ziploc bags to minimize evaporation. Seeds were stratified either in a refrigerator “butter keeper” at 4°C or a wine refrigerator at about 14°C. The seeds in the butter keeper were moved to the wine refrigerator after two months because they were not germinating. When germination was observed, the pots or petri dishes were placed under artificial lighting at room temperature.

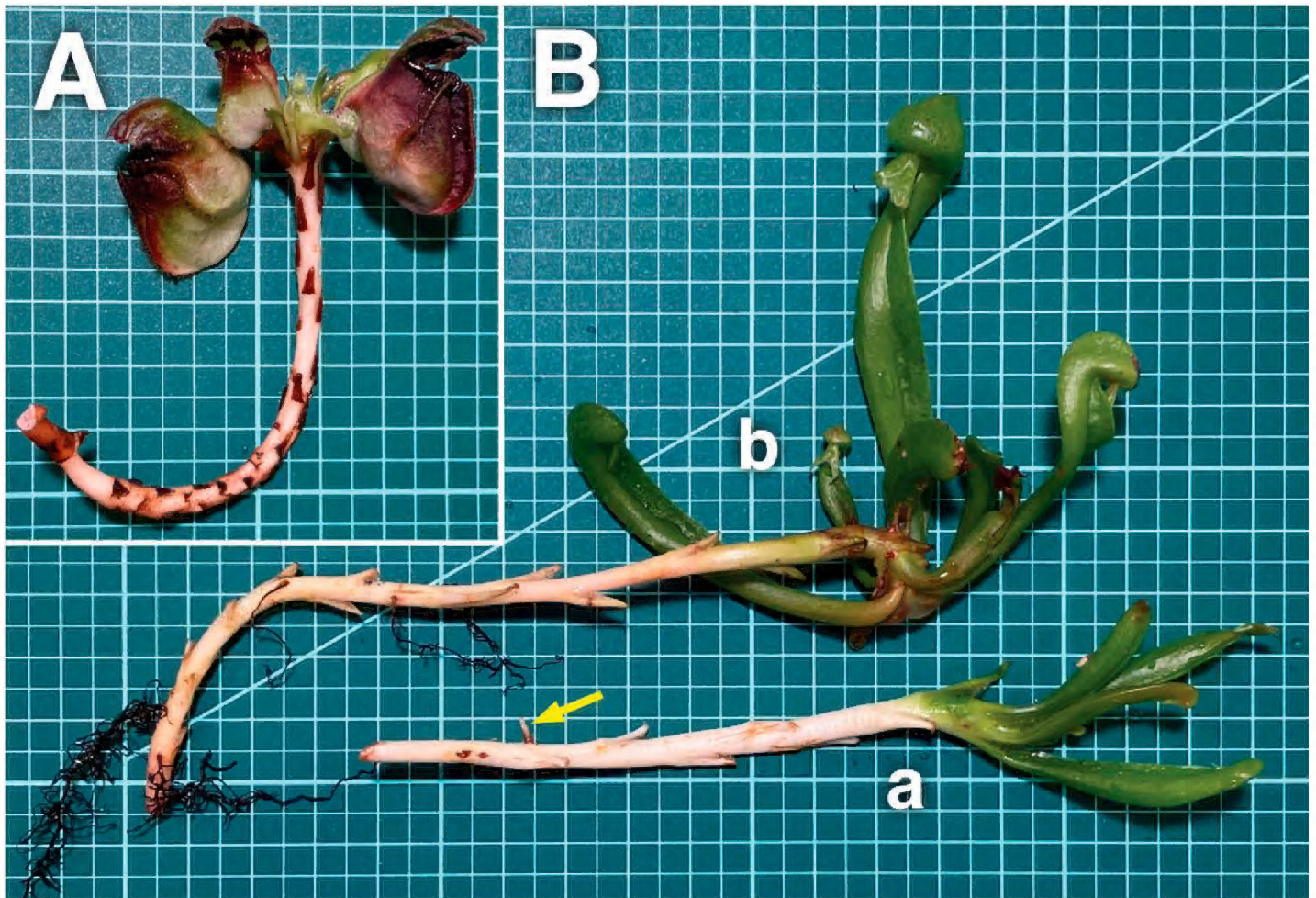


Figure 1: A: Excavated *Cephalotus* stolon with plant that will form a vertical stem at the tip. B: *Darlingtonia* stolons with plants at the tip that will become rhizomatous. Yellow arrow indicates emerging root on stolon. Photographed on a 5 mm grid.

Terminology

The common term for the underground stem-like parts of *Cephalotus* is “rhizome” (c.f. Cross *et al.* 2019). This usage is understandable for instances where herbarium sheets are used to describe a plant and the presence of roots is used to distinguish rhizomes from stolons. However, this usage is misleading in that it mischaracterizes how the plant actually grows and vegetatively reproduces. The root distinction will not be used here.

Cephalotus does not creep along the ground like *Sarracenia*, which unquestionably has rhizomes. Instead, single growth point plants from seed have a short vertical, above ground stem and a tap root. Multi growth point plants are more complicated. Mature plants spread and reproduce vegetatively via subterranean stolons that are typical of other plants that send out stolons on or just below the soil surface. That is, the plants send out elongated stem-like shoots with radial symmetry for the purpose of asexual reproduction. *Darlingtonia* is an example of a plant that sends out stolons and has rhizomes at the stolon tips. In Figure 1Ba, the stolon is in its second year of growth with new leaves at the tip and a few emerging roots. The first year there was no indication of a plant at the tip. Figure 1Bb shows a stolon in its third year with a more mature plant and obvious roots along the stolon. My observation based on digging up stolons and dividing plants is the stolons typically send out roots after the tip starts to become rhizomatous. However, it is quite possible to find stolons doing the unexpected such as a few small pitchers among the scale-like leaves in the middle of a very long stolon. *Cephalotus* stolons produce new plants with vertical stems when they reach the soil surface (Fig. 1A). As the stolons age, they change form and become stolon tubers which

produce roots and additional stolons as shown below. *Cephalotus* stolons can produce roots directly as in the case of propagation by stolon cuttings and can naturally branch as reported by Lowrie (2014) and Cross *et al.* (2019).

Flower and seed observations

The basic description of flowers with the phases of flowering in *Cephalotus* are outlined in detail in Cross *et al.* (2019). The observations reported here provide a more detailed timeline.

Cephalotus has rather simple flowers. Typical flowers have 6 tepals, 12 stamens, 6 stigmas and produce up to 6 seeds (Fig. 2). Flowers open in late spring and summer. On the day flowers open (day 1), by midafternoon, there is shed pollen evident on many of the anthers, but the stigmas have not matured. Additional anthers shed pollen on days 2 and 3. The stigmas start to separate and mature on day 4. The anthers turn brown on about day 9 (range 6–12, n=7), and the flowers start to close and nod down.

Cephalotus pollen is very powdery. It does not stick to small inexpensive natural hair watercolor brushes that work well to pollinate most *Sarracenia* flowers. If the flowers are not disturbed until about day 5, a brush can be used to dislodge a small cloud of pollen from the anthers to self-pollinate a flower. Plucking pollen-laden stamens with a needle nosed forceps and dusting the stigmas of an older flower may work if it can be done gently enough. House flies (Muscidae) and flower flies (Syrphidae) were observed visiting flowers during this study and interfered with observations.

The achenes with seeds dry and splay out about day 30 (range 28–33, n=3) (back cover). An achene with the seed is very light and the long hairs allow the structure to blow in the slightest breeze (Fig. 3A). When closely handling the achenes, it is advised to wear a face mask to keep from breathing on the achenes and scattering them. Just *et al.* (2019) consider that the seeds with achenes are primarily wind dispersed. They will also float on water, which would be advantageous to both dispersal and placement at higher locations in a bog during seasonal flooding, as was reported by DeBuhr (1976) and like what was observed for *Drosera* species (Ksepka 2019; Bourke 2021). I have observed that the achenes are strikingly similar to *Darlingtonia* seeds and could be dispersed long distance by clinging to the fur of an animal visiting nearby exposed water as described in Collingsworth (2015) for *Darlingtonia*. *Cephalotus* does not have all the seed dispersal mechanisms of *Darlingtonia*. The most significant is *Darlingtonia* can fling seeds onto a host (personal observation). *Darlingtonia*, like *Cephalotus*, has very tall flower stalks that can swing when disturbed. In the field, just brushing past a mature *Darlingtonia* seed capsule will result in seeds being thrown



Figure 2: *Cephalotus* ‘OG Black’ flowers. Note some flowers have unusual numbers of tepals and stigmas.

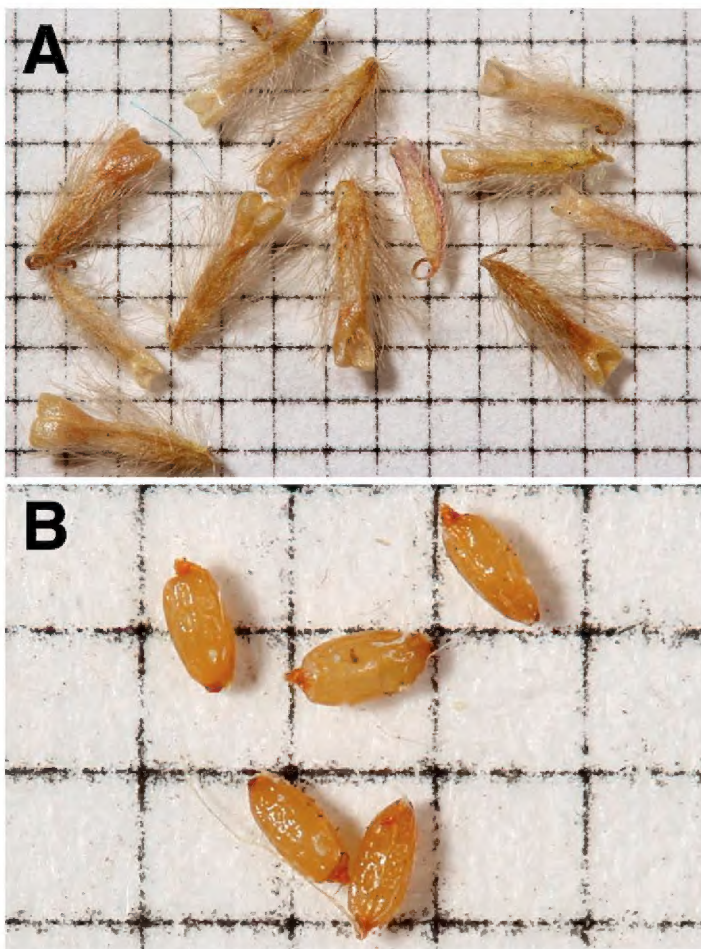


Figure 3: *Cephalotus* achenes (A) and seeds (B) photographed on a 1 mm grid.

Because some of those achenes were collected with forceps which can eject seeds from the achenes and forceps can damage seeds as well, the trial was repeated in 2022 collecting seeds carefully into a large bowl. No free seeds were found in the bowl after collection. In this sample, only 59 seeds were collected from 197 achenes (30% contained seeds). The conditions the plants were growing under were not natural, but this general observation could account for the results of Just *et al.* (2019) that they had a substantially lower rate of seedling success when the seeds were not removed from the achenes. Under natural conditions there is an advantage for the seeds to remain in the achene. The achene is more likely to remain on the surface of the soil. As seen in the next section, seedling development is most amenable to being on the surface of the soil.

Seedling development

Just *et al.* (2019) studied *Cephalotus* seed germination in the context of its ecology and conservation biology. They did extensive studies of seed storage and germination requirements. As part of their study, they determined the best conditions to induce seed germination. The most important factors were to hydrate the seeds and to stratify them at 15°C. This is not “cold stratification” in the usual sense. As they found and I reproduced, stratifying seeds colder was not helpful and they would not germinate properly until moved to about 15°C. They needed to be at 15°C about 6 to 10 weeks. Just *et al.* (2019) found the seeds that did not germinate properly were not necessarily dead. They could generally be coaxed to germinate with longer stratification at 15°C or with the hormone GA3. Figure 4 displays the terminology used in describing seedlings.

Cephalotus seeds appear not to contain small embryonic plantlets as are typical in common flowering plants. Instead, it appears there is simply a mass of undifferentiated embryonic cells from

out of the capsule. This is very advantageous for *Darlingtonia* because the plants send out large numbers of stolons that can completely colonize all the immediate habitat capable of supporting the plants in a few years to decades depending on the site. There is a distinct advantage for the seeds to go to other wetlands in the area as there would be few opportunities in the immediate vicinity. *Cephalotus* flower stalks do not hold the individual seeds very long but do mature seeds daily for about two months. In addition, the stolons that *Cephalotus* plants produce tend not to make massive long term stands in their usual habitat (Nunn 2014). Hitching rides on animals should be possible for less common but important longer distance dispersal between bogs.

Cephalotus sometimes produces achenes without seeds. Note in Figure 3A that at least four of the achenes appear not to be large enough to contain a seed (Fig. 3B). In a sample of achenes from this study in 2021, 44 seeds were collected from 94 achenes (47% con-

which the plantlet structures differentiate and grow after germination, defining germination as when the seed expands and the embryonic tissue starts to extrude (Fig. 5A-B). Cross *et al.* (2019) have a photo of a longitudinal section of a seed. They interpret the photo to show a small white linear embryo at the distal tip of the seed surrounded by endosperm. It is presumed during stratification the endosperm breaks down nourishing the embryo so that at the time of germination, the remains of the endosperm become the membrane-like structure enclosing the embryo which at that time fills the seed. This process needs more study because at the beginning of germination, there is no outward evidence of any differentiation of the embryo. Instead, it appears that as the undifferentiated embryonic cells expand at germination and the seed coat splits, an opening occurs at the distal end of the enclosing membrane, and the embryonic cells are extruded (Fig. 5B-C). It is not clear how much of this extrusion process is powered from the embryonic cells expanding and how much is from hidden differentiation and development of the hypocotyl and later, the cotyledons within the seed coat and membrane (Figs. 5C-E, 6A-B). It is unclear how the cotyledons get out of the enclosing membrane because it appears intact after cotyledons are out. Conran & Denton (1996) refer to the embryonic cells as a hypocotyl extension, but developmentally it appears the hypocotyl sprouts from the cells.

The result of this delayed differentiation is the immediate development of a seedling substantially larger than the seed (Fig. 5E, 6B). Most seedlings maintain the mass of embryonic cells between the hypocotyl and radicle (Fig. 6C). A few seedlings were observed that did not have an obvious mass of undifferentiated cells (left side of Fig. 5E). These seedlings were much smaller. After the seedlings exit the seeds, it appears the embryonic cells do not provide much substance to the seedlings and may only play a developmental role that is complete when the seedling is complete. Figure 6D shows a seedling six months after being planted out. The blue arrow points to the embryonic cells that have become a knob at the hypocotyl-radicle junction. The embryonic cells and hypocotyl appear to have become a typical stem and the radicle a fleshy stem-like tap root. Note the typical adventitious roots pointed to by the yellow arrows.

Jan Schlauer (pers. comm.) suggested that what here are called undifferentiated embryonic cells could be a tuber-like storage structure formed from the primary root, and the observed root is a secondary, adventitious root. This is an interesting hypothesis that offers a more traditional interpretation of what is seen in the seedlings. Figure 7 shows more of the variation among the seedlings. There are seedlings with the hypocotyl on the proximal end of the cell mass and the “radicle” on the distal end. However, most seedlings have both the hypocotyl and radicle attached laterally to the mass after it is out of the seed coat. If the shape of the cell mass does not reflect the prior shape inside the seed and the hypocotyl and cotyledons existed before germination, the tuber hypothesis

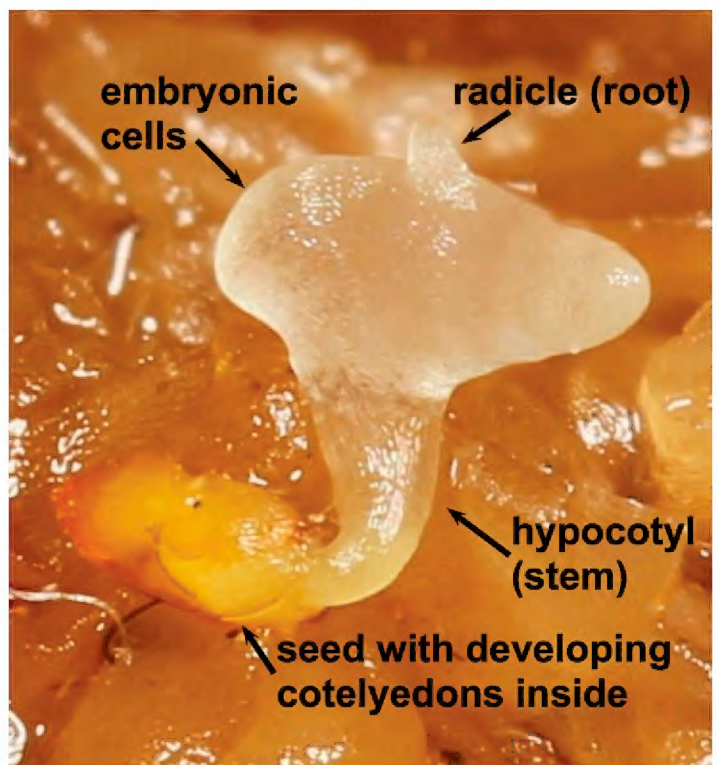


Figure 4: *Cephalotus* seedling emerging from seed. It appears the seed initially just contains embryonic cells. The hypocotyl and radicle sprout from the cell mass as the seedling develops at germination.

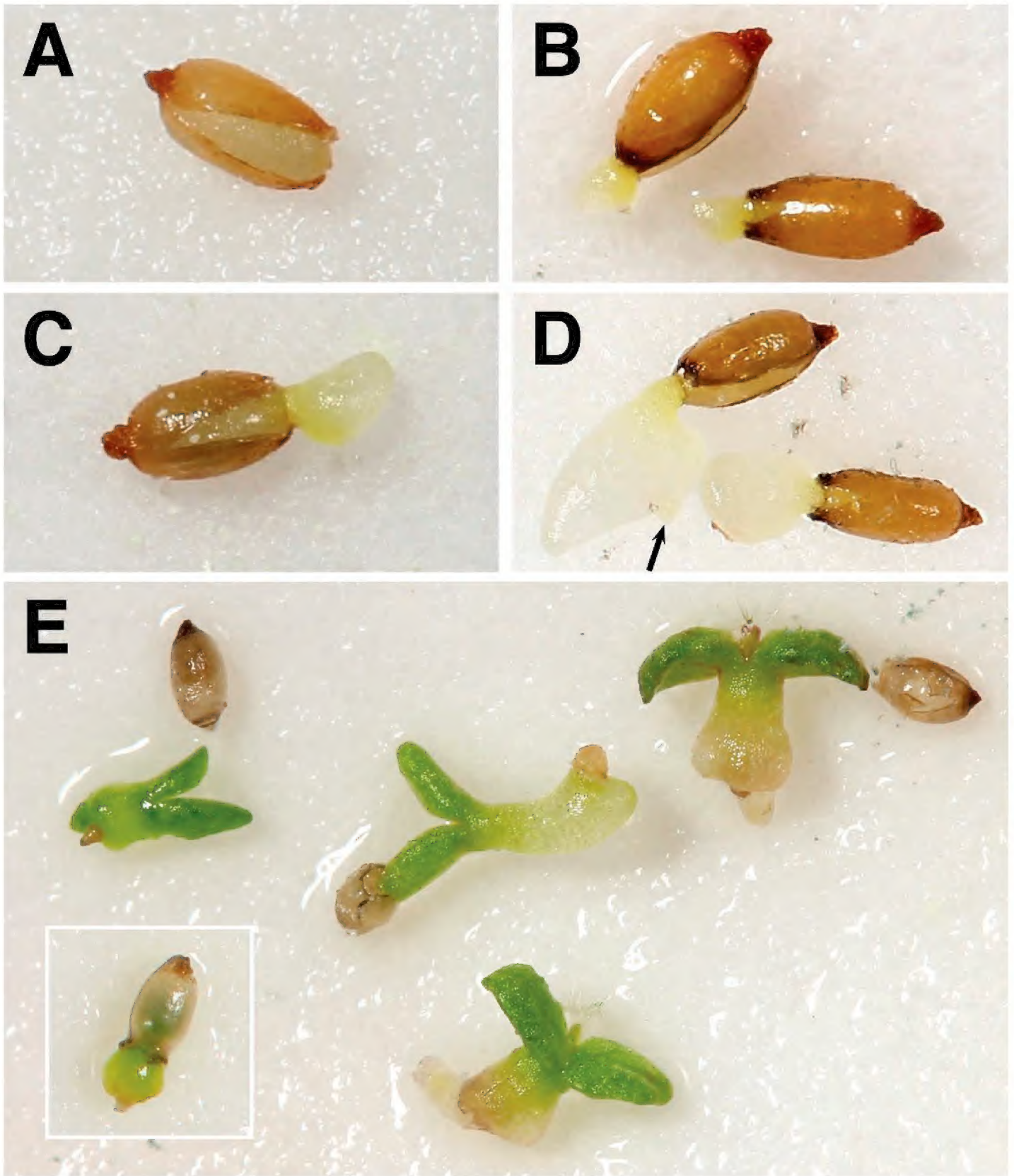


Figure 5: *Cephalotus* stages in seedling development. A: Embryo swells and seed coat splits. B-C: Embryonic cells continue to swell and extrude out of the enclosing membrane. D: Embryonic cells are out of the seed membrane. The hypocotyl is beginning to emerge. Arrow indicates differentiating root. E: Fully developed and emerged seedlings. Inset was taken 15 days earlier showing cotyledons still within seed membrane. A-D were in darkness. E was arranged with seed coats for scale—the upper left seed coat may not go with that seedling. Inset may not be the same seedling as above it.

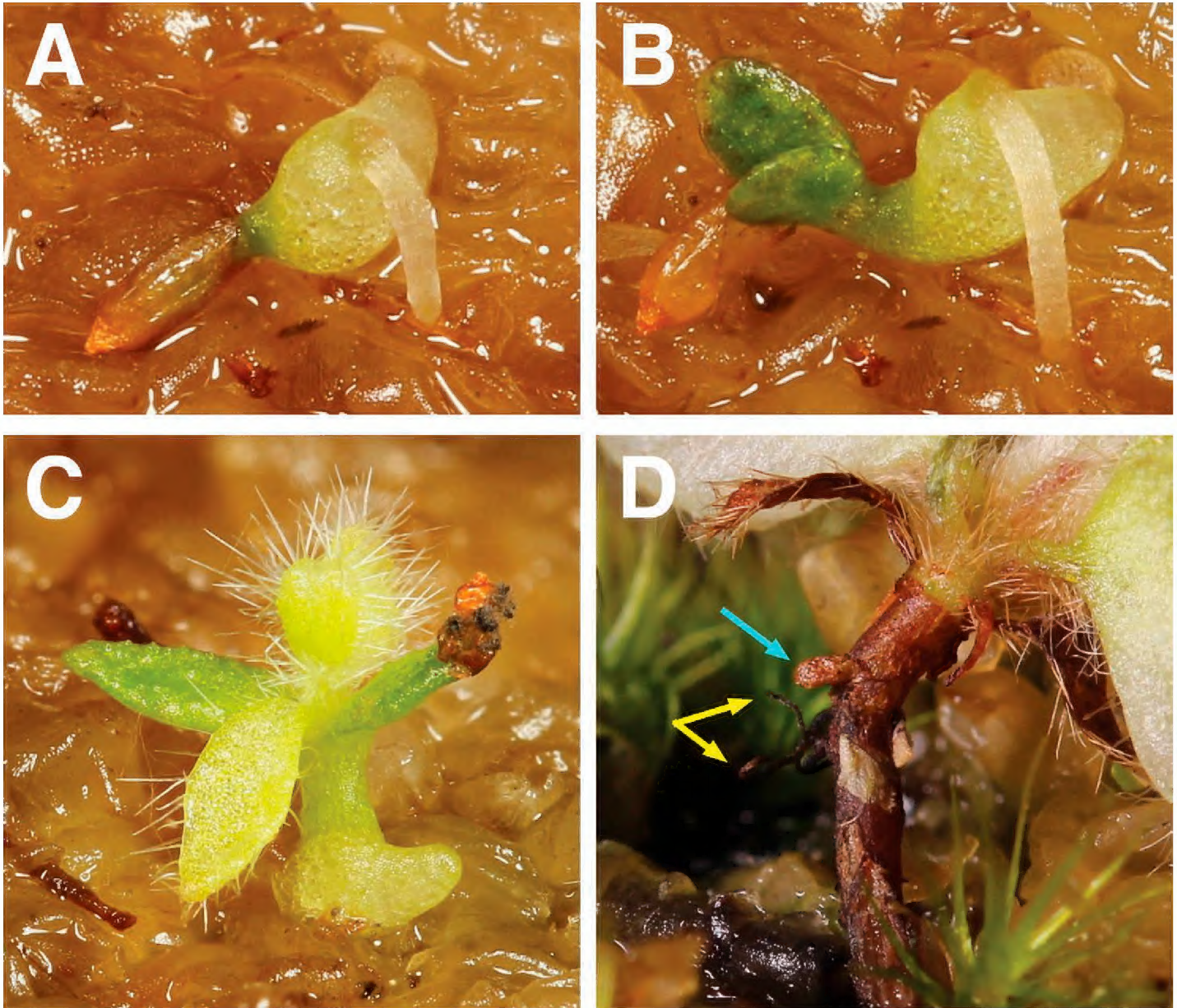


Figure 6: *Cephalotus* advanced stages in seedling development. A-B: Embryo with radicle entering the soil before and after release of cotyledons from the seed coat. C: Planted seedling showing the first leaf which is a foliage leaf and the second leaf which is a pitcher. This is typical. D: Partly excavated seedling six months after planting. The blue arrow points to the remains of the embryonic cells at the hypocotyl-radicle junction. The yellow arrows point to typical *Cephalotus* adventitious roots emanating from the radicle (tap root).

would be possible. To determine how typical or unique *Cephalotus* germination is, it would require a classical 19th century microscope study of seed development and germination.

In the wild, *Cephalotus* seeds should germinate during the cool, rainy winter. Six months later they will be presented with a warm to hot, dry summer. To survive the summer, the seedlings need moisture. Having relatively large seedlings and a fleshy tap root for water and nutrient storage could be critical for the seedling survival on the drier edges of bogs and seasonally variable seeps where they are found.

Reproduction via stolons

According to my observations from propagating and growing *Cephalotus*, it appears that the plants immediately from seed have short taproots that ultimately become thickened and tuberous. The tubers bud off adventitious roots (Fig. 6D). Older plants from seed spread via stolons. These stolons form short leafy vertical stems at the soil surface.



Figure 7: *Cephalotus* seedlings showing the variation found. The same seedlings may appear more than once but at different growth stages.

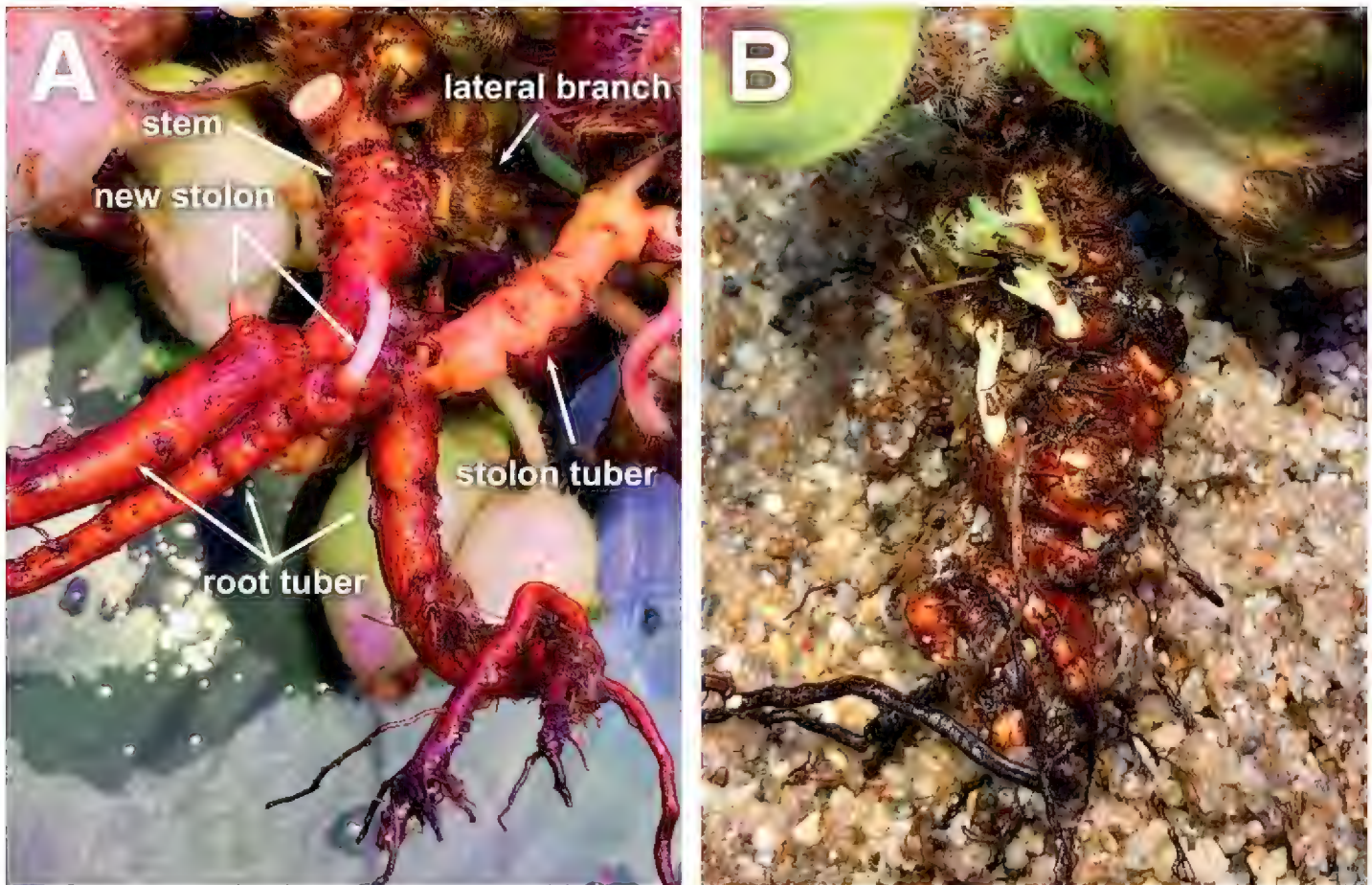


Figure 8: A: *Cephalotus* 'OG Black' plant two years after propagation via division. New stolons are originating on root tubers. An older stolon is tuberous. Lateral branches off the main stem tend to be short lived. B: *Cephalotus* (from Denbarker, WA seed) plant two years after propagation via stem cutting. Stolons are sprouting from the stem after the main growth point died post blooming. The underground part of the stem has become knobby and tuber-like. This clone tends not to be as prolific at producing root tubers compared to the clone in A.

The above ground stems of *Cephalotus* are usually no longer than a few cm. Growth tips die when the plant blooms or, if the plant does not get the appropriate light cues to bloom, just dies with age. This event usually initiates the production of stolons and lateral stem branches (Fig. 8).

The subterranean parts of mature *Cephalotus* plants are typically a tangled mass of stolons, roots, and buried stems. Most of the stolons and some of the roots are in the form of tubers. It can be difficult to tell the stolon tubers from the root tubers (Fig. 8A). Buried stems become knobby and somewhat tuberous (Fig. 8B). New stolons sprout from the stolon tubers, root tubers, and buried stems. Cross *et al.* (2019) have a photo of a cross section of a tuber showing a mass of parenchyma cells around the original root or rhizome. Having relatively large storage and freely reproducing subterranean structures allows *Cephalotus* plants to quickly recover after bush fires (Nunn 2014). They also make the plant easy to propagate in cultivation.

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**PROBOSCIDEA AND DICYPHINI: AN EXAMPLE OF SPONTANEOUS MUTUALISM
BY A POPULATION OF PREDATORY BUGS INHABITING A STICKY PLANT**

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Keywords: *Dicyphus errans*, Miridae, mutualism, *Proboscidea*, *Ibicella*, Martyniaceae, Byblidaceae, Droseraceae, Roridulaceae, arthropods, Devil's Claw.

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Abstract: A population of the mirid-bug *Dicyphus errans* (Miridae) a species occurring in Europe, colonized a group of Devil's Claw (*Proboscidea louisianica* subsp. *fragrans* (Martyniaceae)) native to the USA and cultivated in a garden in Germany. More than 20 individuals of these predatory bugs have been found living and moving freely on these sticky plants, feeding on stuck victims, while garden ants (*Lasius* spp. (Hymenoptera)) trying to place aphids for "sugar farming" became captured together with their "productive livestock", as do small flies and bees. The observations on this spontaneous mutualism between the American Devil's Claw and a small mosquito-like European mirid-bug has been recorded on video (Hartmeyer 2022). This constitutes an additional example for the adaptation of these ubiquitous predatory bugs to feed on the prey of sticky plants that would usually capture small-sized insects. The American Frank Obregon (2017), who found native mirid-bugs on his cultivated *Roridula* (Roridulaceae) in California, called the phenomenon assisted carnivory, which fits quite nicely. *Proboscidea* as well as *Roridula* do not produce digestive enzymes, so they cannot benefit directly from their diverse captured prey. But as soon as predatory bugs colonize the plants and feed on the sticking prey, their droppings provide a suitable fertilizer to be absorbed by the stomata of the leaf. A perfect mutualism. In addition, some historic facts on bug-plant interaction are provided.

A short history on Miridae and the designation "assassin bugs"

The Assassins (also called Hashashins), were notorious for attacks and murder with dirks and poisoning during the 11th to 13th century in Persia and Syria. The terms "assassin" and "assassination" go back to this group as a synonym for killer or murderer and murder, respectively. They were first introduced to the Italian and later to the English language. The trivial names assassin bug or heath assassin bug have been assigned to the predatory true bug *Coranus subapterus* (Reduviidae) (De Geer 1773). Furthermore, "assassin bugs" became a common name for the family Reduviidae, because these large predators use their pronounced proboscis very violently to stab prey to death, as the Assassins were said to do.

In the same manner as it became usual in the English language to call all insects "bugs", "assassin bug" became an umbrella term for all predatory true bugs. This is only marginal if used colloquially; it is, however, not fit to scientific standards in publications. Let us take a glance at the history. In the early 20th century, Francis E. Lloyd learned from A. G. Hamilton, that *Byblis gigantea* harbors a small insect which he called a "buttner". Later, H. Stedman provided the same information and guided Lloyd to the natural habitat. In his book "The Carnivorous Plants", Lloyd (1942) reported for

the first time about the Australian mutualistic bugs, using the term “capsid” derived from the family name Capsidae (Hemiptera): “At some distance north of Perth we found a lot of plants ... infested with a small wingless capsid which turns out to be a new genus and will be described by Dr. W. R. China of the British Museum”. Furthermore “Similar insects, also capsids move freely without difficulty over the surface of *Drosera* leaves in Australia, and of the African genus *Roridula*”. Indeed, Dr. China described the mentioned capsid bugs found in the surroundings of the town Perth (China & Carvalho 1951; China 1953). However, in those days it was a rather exotic issue.

In the 1980s, the public interest in carnivorous plants increased significantly and subsequently also the availability of artificially propagated CPs. Particularly in Western Europe, *Roridula* plants became available, including their mutualistic *Pameridea* bugs (Miridae). In the 1990s, the authors travelled over months through Australia to study and film carnivorous plants. Already experienced for several years in the cultivation of *Roridula* and *Pameridea* in Germany, in 1995 we stumbled coincidentally upon several new *Setocoris* (Miridae) and *Cyrtopeltis* species (Miridae) living on *Byblis filifolia* (Byblidaceae) and various sundew species (*Drosera*). It was the first documentation far away from the former known records in the Southwest, on widespread plants in remote tropical regions of Western Australia and in the Northern Territory (Hartmeyer 1996 and videos Hartmeyer & Hartmeyer 1995; Hartmeyer 2016).

In 1995, when translating the documentary into English, the authors realized that the general use of the umbrella term “assassin bugs” for capsids leads to misinterpretations. As a result, the problem was first discussed in the Bulletin of the Australian Carnivorous Plant Society (Hartmeyer 1996) with regard to the newly found tropical mutualisms: “The comprehensive term assassin bugs – up to now current – should however not be used since this name is already given to the bug *Coranus subapterus*.” In the comprehensive book Carnivorous Plants of Australia Magnum Opus (Lowrie 2014), the author who found even more plant-bug associations in tropical Australia in the late 1990s, quoted Hartmeyer’s sentence and suggested to replace the misleading term “assassin bugs” by “sundew bugs”. This sounds good and is probably less misleading. But as these bugs are found on *Byblis* as well, the term would be inappropriately exclusive. Therefore, it is recommended to use simply the family names Miridae, mirids or mirid bugs, respectively the synonym Capsidae, capsids or capsid bugs. In recent literature, Miridae is used predominantly.

Garden ants and domestic mirid bugs find a cultivated *Proboscidea louisianica*.

In late March 2022, the sown seeds of *Proboscidea louisianica* subsp. *fragrans* (Lindl.) P.K. Bretting (syn. *P. fragrans* (Lindl.) Decne.) germinated inside our greenhouse and developed nicely. At the end of April, it was warm enough to move the now already flowering plants into our garden. It needed only a day and the sweet aromatic scent of the pretty flowers attracted garden ants, which immediately tried to place aphids on the sticky plants. But they were unlucky! The ants, as well as the aphids, stuck to the numerous sticky hairs of the plant (Fig. 1), no longer able to free themselves. After the first week the *Proboscidea* looked typical: Particularly the lower leaves were covered with dead ants, aphids as well as small flies and bees. On a first glance, also mosquitos seemed to stick on the leaves; however, a closer look showed that they walked freely over the plant. That seemed very unusual and a macro-shot brought to light that they were mosquito-like domestic mirids of the genus *Dicyphus* (Fig. 2). What a nice surprise! There are several closely related species, only distinguished by the shape of their penis, which we did not examine. However, most likely a population of the predatory bug *Dicyphus errans*, specialized to hunt on sticky plants and mulleins, had settled on all six of our *Proboscidea* plants. An ideal set to record the mutualism on video.



Figure 1: *Lasius* ants: A) Attracted by a fallen flower; B) Captured by the sticky plant.

Four weeks later, more than 20 bugs lived on four flowering plants inside a 34 cm pot (Fig. 3), placed on a garden table, providing a good view around. When one approached the plants, the mirids hid on the lower leaf side but did not fly away. After growing *Roridula* with *Pameridea* bugs for many years, the scenery looks now very similar, and regarding the mutualism, we observed just another example of assisted carnivory. Frank Obregon (2017) had a similar experience when domestic mirids settled on his cultivated *Roridula*. If sticky plants, unable to produce their own digestive enzymes, capture diverse prey, they cannot make use of it directly. But as soon as mirid bugs assist in digesting the prey and leave their droppings on the leaves, the prey nutrients may become available for the plant too. A clear win-win situation, which is obviously more widespread in nature than generally known.

Such assisted carnivory is at least a part of the digestion in almost all known plants acknowledged as truly carnivorous (Hartmeyer 1997). Different organisms even build complex food webs in pitchers of many truly carnivorous *Nepenthes* species (Clarke 1997). In this regard, it is important that most of the commensal mirids are associated with non-carnivorous plants: *Pinalitus parvulus* is connected to *Salvia canariensis*, *Apolygus lucorum* to *Passiflora* species, and *Dicyphus agilis* and *Nesidiocoris tenuis* to *Nicotiana tabacum* (tobacco) and *Solanum* (tomatoes and potatoes). Recently Miridae even became interesting as biological pest control for associated plants (Martinez et al. 2014). Associated bugs feed on parasites and victims that stick coincidentally to the defensive glands and they leave their droppings on the host plants.

If the term assisted carnivory were accepted to turn *Roridula* and *Proboscidea* into true carnivores, it would – if no clear limitations were defined – also turn tobacco, tomatoes, and potatoes into true carnivores. Generally, it does not matter if the bugs are present for a lifetime or temporarily after plants used volatiles to “announce” parasite-attacks (observed in *Nicotiana*) to their “partners”. Without any limiting definition for the term assisted carnivory, all those herbs would become at least temporary true carnivores. Even worse, is it true carnivory when herbs benefit from nutrients broken down by maggots that fed on dead animals on a meadow? Carnivorous grass? Discussion opened! To provide a sufficient answer now would certainly exceed the scope of this article.

Pleasant feedback on the first go

Only one day after uploading our video recordings on YouTube on 16 June 2022 (Hartmeyer 2022), combined with the request to report bug colonization on own Martyniaceae, we received several reports with photos via Facebook and e-mail. These confirmed the described mutualism



Figure 2: *Dicyphus errans*: A) Freshly hatched nymph; B) Nymph feeding on prey; C) Unwinged nymph after 1st moult; D) Winged nymph moulting; E) Empty exoskeleton after moult; F) Adult.

at the first go for two more European countries. *Dicyphus errans* on *Proboscidea* or *Ibicella* was found by Nils Klare (West Germany), Constantin Dormann (West Germany), Mathias Maier (South Germany, already for five years), Stéphane Joly (North France), and Jure Slatner in Slovenia. Jure had the bugs on his *Proboscidea* determined as *Dicyphus errans* by a Slovenian expert and reported



Figure 3: *Proboscidea louisianica* subsp. *fragrans* with *Dicyphus errans* population.

about it in a Slovenian book he wrote (Slatner 2019). In fact, he posted photos of the settlement in a Slovenian CP-forum as early as December 2008. Unfortunately, it was probably due to the language barrier that these early observations were not further examined. Remarkably, all photos sent to us, from northern France through Germany to Slovenia show identical bugs.

From the U.S., Matt Paddock mentioned a natural site in California with wild growing *Proboscidea louisianica* that seem to host a few different species of Miridae, very similar to our photos. Many of the mirids are not host specific, he wrote. That is certainly true. We also find *D. errans* on mullein in our garden; however, with fewer individuals than on the *Proboscidea*. Although an American plant is colonized by European bugs here, the wide distribution of this spontaneous mutualism confirms both partners are obviously well matched.

Dr. Barry Rice (2008) reported in CPN a mutualism in California. He found (probably) *Cyrtopeltis modesta* (Miridae) on *Ibicella lutea* (Martyniaceae). So, it doesn't matter where these Martyniaceae grow, they turn out to be typical bug plants and usually find partners. Their choice is quite impressive, as in Europe alone >1,200 species of Miridae are known. Some more papers (Anderson & Midgley 2002; Ellis & Midgley 1996; Heslopp-Harrison 1976; Penn State New Kensington 2014; Lloyd 1934; Voigt & Gorb 2008) add to the topic.

Finally, a look at the scene in our garden in the far southwest of Germany. The plants have developed well during a hot summer until early August and bear fruit abundantly by now (Fig. 3). On 25 June 2022, I found the first newly hatched nymphs of *D. errans* (Fig. 2), moving over sticky leaves and stems as easily as the adults. They found plenty of food and moulted after about 1-2 weeks. Only after the 2nd or 3rd moult, around mid July, the bright green nymphs with their striking red eyes formed wings for the first time. So far plants and bugs are doing well.

Methods: Photography and videography were done using an Olympus SH2 for photos and a Sony HXR-NX 80 with Zeiss Vario-Sonnar lens for 4K-video.

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THE TAXONOMIC RELEVANCE OF NAPHTHOQUINONES IN TROPICAL PITCHER PLANTS (*NEPENTHES* L., NEPENTHACEAE)

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Abstract: The distribution of the naphthoquinones ramentaceone and plumbagin was studied among 50 taxa of the genus *Nepenthes*. Naphthoquinone patterns support classifications based on homology of plastid and/or nuclear genes to some extent, with plumbagin predominant in sections *Nepenthes*, *Urceolatae*, *Tentaculatae*, and *Regiae*, ramentaceone predominant in sections *Insignes* and *Villosae*, and both isomers present without clear predominance in sections *Pyrophytae* and *Montanae*. Only 9 of 96 studied species contained both isomers in the same plant. Naphthoquinone data from artificial hybrids of known parentage allowed conclusions on the biosynthesis of these compounds and the heredity of the respective enzymatic steps.

Introduction

The taxonomic utility of the distribution of the naphthoquinone isomers ramentaceone (7-methyljuglone, **M**) and plumbagin (2-methyljuglone, **P**) in the genus *Nepenthes* L. (Nepenthaceae) was already indicated by the first systematic screening (Schlauer *et al.* 2005). In some respects, the distribution of the isomers throughout the genus resembles patterns found in the closely related family Droseraceae (Culham & Gornall 1994; Schlauer *et al.* 2017; Schlauer *et al.* 2018; Schlauer *et al.* 2019a-c; Schlauer & Fleischmann 2021; Schlauer & Fleischmann 2022). All *Nepenthes* species investigated so far share the same chromosome count of $2n = 80$ (Heubl & Wistuba 1998), while chromosome counts in Droseraceae are more diverse, with most species (ca. 53%) having $2n = 20$ chromosomes or counts based on the base number $x = 10$ (followed by species with counts based on $x = 7 \approx 18\%$ and $x = 6 \approx 16\%$, Kondo & Lavarack 1984; Kondo & Segawa 1988; Hoshi & Kondo 1998; James *et al.* 1997; Rivadavia 2005), so *Nepenthes* may be polyploid (octoploid with respect to a base count of $x = 10$). Together with the fact that Nepenthaceae are dioecious (obligately outcrossing) and genetically compatible across species boundaries, a comparatively high degree of hybridity can be expected for most taxa, especially where several different ones co-occur (Scharmann *et al.* 2021). Nevertheless, most *Nepenthes* species contain just one of the possible two naphthoquinone isomers (Schlauer *et al.* 2005). Taxa containing both isomers can thus be assumed to be of comparatively recent hybrid origin, similar to the situation in *Drosera* (Schlauer & Fleischmann 2016).

As naphthoquinone formation is widespread among Nepenthes, species devoid of naphthoquinones are most probably derived from ancestors that produced them. Hybrids of naphthoquinone-free species with naphthoquinone producing species can be expected to contain the quinone



Figure 1: 1 = *N. ampullaria*, 2 = *N. inermis*, 3 = *N. ventricosa*, 4 = *N. talangensis*, 5 = *N. reinwardtiana*, 6 = *N. stenophylla*, 7 = *N. eymae* x *clipeata*, 8 = *N. rafflesiana*. Photos: S. Hartmeyer.

Table 1. <i>Nepenthes</i> taxa investigated and quinones found.				
Section ^a	Taxon	Provenance/comment ^b	Quinone(s) ^c	Ref. ^d
<i>Nepenthes</i>	<i>N. danseri</i>	New Guinea	P	1
<i>Nepenthes</i>	<i>N. distillatoria</i>	Sri Lanka	P	1
<i>Nepenthes</i>	<i>N. khasiana</i>	N-India; (SH)	P	*, 1
<i>Nepenthes</i>	<i>N. lamii</i>	New Guinea (Doorman Top)	P	1
<i>Nepenthes</i>	<i>N. madagascariensis</i>	Madagascar	P	1
<i>Nepenthes</i>	<i>N. masoalensis</i>	Madagascar	P	1
<i>Nepenthes</i>	<i>N. neoguineensis</i>	New Guinea; (AW)	P	*, 1
<i>Nepenthes</i>	<i>N. paniculata</i>	New Guinea; (AW)	P	*
<i>Nepenthes</i>	<i>N. pervillei</i>	Seychelles (Mahé, Morne Seychelloise); (SH)	P+M	*, 1
<i>Nepenthes</i>	<i>N. tomoriana</i>	New Guinea	P	1
<i>Nepenthes</i>	<i>N. treubiana</i>	New Guinea; (AW)	P+M	*
<i>Nepenthes</i>	<i>N. vieillardii</i>	New Caledonia	0	1
<i>Urceolatae</i>	<i>N. ampullaria</i>	Sumatra	0	1
<i>Urceolatae</i>	<i>N. bicalcarata</i>	Borneo (Malaysia)	P	1
<i>Urceolatae?</i>	<i>N. gracilis</i>	cult.	P	1, 4
<i>Urceolatae?</i>	<i>N. hirsuta</i>	Borneo	0	1
<i>Urceolatae?</i>	<i>N. mapuluensis</i>	Borneo	0	1
<i>Urceolatae?</i>	<i>N. mirabilis</i>	cult.	P	1
<i>Urceolatae?</i>	<i>N. papuana</i>	New Guinea (Doorman Top)	P	1
<i>Tentaculatae</i>	<i>N. glabrata</i>	Sulawesi	P	1
<i>Tentaculatae</i>	<i>N. hamata</i>	Sulawesi	P	1
<i>Tentaculatae</i>	<i>N. muluensis</i>	Borneo (Mulu)	P	1
<i>Tentaculatae</i>	<i>N. tentaculata</i>	Sulawesi & E Borneo	P	1
<i>Tentaculatae</i>	<i>N. undulatifolia</i>	Sulawesi; (AW)	P	*
<i>Insignes</i>	<i>N. bellii</i>	Philippines (Mindanao)	P+M	1
<i>Insignes</i>	<i>N. biak</i>	New Guinea (Biak); (AW)	M	*
<i>Insignes</i>	<i>N. burkei</i>	Philippines	M	1
<i>Insignes</i>	<i>N. campanulata</i>	Borneo	0	1
<i>Insignes</i>	<i>N. insignis</i>	New Guinea (2 accessions: Biak & type loc.) (AW)	M	*, 1; P in 2
<i>Insignes</i>	<i>N. merrilliana</i>	Philippines (Mindanao); (SH)	0	*, 1
<i>Insignes</i>	<i>N. sibuyanensis</i>	Philippines (Sibuyan); (AW)	M	*, 1
<i>Insignes</i>	<i>N. ventricosa</i>	Philippines (“green”)	M	1
<i>Insignes</i>	<i>N. ventricosa</i>	Philippines; (“porcelain”); (SH)	P	*, 1
<i>Villosae</i>	<i>N. boschiana</i>	Borneo	P	1
<i>Villosae</i>	<i>N. copelandii</i>	Philippines (Mt. Apo)	M	1
<i>Villosae</i>	<i>N. edwardsiana</i>	Borneo; (AW)	M	*

Table 1. Continued.				
Section ^a	Taxon	Provenance/comment ^b	Quinone(s) ^c	Ref. ^d
<i>Villosae</i>	<i>N. macrophylla</i>	Borneo (Trus Madi)	M	1
<i>Villosae</i>	<i>N. micramphora</i>	Philippines (Mindanao); (AW)	P	*
<i>Villosae</i>	<i>N. pulchra</i>	Philippines, (possibly hybrid with another species) (SH)	M	*
<i>Villosae</i>	<i>N. rajah</i>	Borneo	P	1
<i>Villosae</i>	<i>N. truncata</i>	Philippines (Mindanao); (SH)	0	*, 1
<i>Villosae</i>	<i>N. villosa</i>	Borneo	P	1
<i>Pyrophytae</i>	<i>N. albomarginata</i>	cult. (“red”); (SH) & Malaysia (Penang)	M	*, 1
<i>Pyrophytae</i>	<i>N. benstonei</i>	Malaysia (peninsular)	P+M	1
<i>Pyrophytae</i>	<i>N. gracillima</i>	Malaysia (Tahan)	P	1
<i>Pyrophytae</i>	<i>N. macfarlanei</i>	Malaysia (peninsular)	P	1
<i>Pyrophytae</i>	<i>N. macrovulgaris</i>	Borneo	0	1
<i>Pyrophytae</i>	<i>N. northiana</i>	Borneo	P	1
<i>Pyrophytae</i>	<i>N. rafflesiana</i>	cult. (SH)	P	*, 1
<i>Pyrophytae</i>	<i>N. ramispina</i>	Borneo; (SH)	0	*, 1
<i>Pyrophytae</i>	<i>N. reinwardtiana</i>	Borneo (“green”); (SH)	P+M	*, 1
<i>Pyrophytae</i>	<i>N. sanguinea</i>	Malaysia (Genting)	M	1
<i>Pyrophytae</i>	<i>N. smilesii</i>	Thailand	P	1
<i>Pyrophytae</i>	<i>N. thorelii</i>	Vietnam	0	1; P in 3
<i>Montanae</i>	<i>N. adnata</i>	Sumatra	M	1
<i>Montanae</i>	<i>N. aristolochioides</i>	Sumatra	P	1
<i>Montanae</i>	<i>N. bongso</i>	Sumatra (Singgalang)	M	1
<i>Montanae</i>	<i>N. densiflora</i>	Sumatra	P+M	1
<i>Montanae</i>	<i>N. diatas</i>	Sumatra (Bandahara)	P	1
<i>Montanae</i>	<i>N. dubia</i>	Sumatra (Barisan)	P	1
<i>Montanae</i>	<i>N. eustachya</i>	Sumatra	P	1
<i>Montanae</i>	<i>N. gymnamphora</i>	Sumatra; (SH)	P	*, 1
<i>Montanae</i>	<i>N. inermis</i>	Sumatra (Gadut)	P	1
<i>Montanae</i>	<i>N. jacquelineae</i>	Sumatra	M	1
<i>Montanae</i>	<i>N. lavicola</i>	Sumatra	0	1
<i>Montanae</i>	<i>N. longifolia</i>	Sumatra	P	1
<i>Montanae</i>	<i>N. miki</i>	Sumatra	P	1
<i>Montanae</i>	<i>N. ovata</i>	Sumatra	M	1
<i>Montanae</i>	<i>N. rhombicaulis</i>	Sumatra	P+M	1
<i>Montanae</i>	<i>N. singalana</i>	Sulawesi (Mantalingajan & Singgalang)	P	1

Table 1. Continued.				
Section ^a	Taxon	Provenance/comment ^b	Quinone(s) ^c	Ref. ^d
<i>Montanae</i>	<i>N. spathulata</i>	Sumatra (G. Tudjuh & Tanggamus)	M	1
<i>Montanae</i>	<i>N. spectabilis</i>	Sumatra (Pangulubao & Bandahara)	0	1
<i>Montanae</i>	<i>N. sumatrana</i>	Sumatra	P	1
<i>Montanae</i>	<i>N. talangensis</i>	Sumatra	P	1
<i>Montanae</i>	<i>N. tenuis</i>	Sumatra	P+M	1
<i>Montanae</i>	<i>N. tobaica</i>	Sumatra (Toba)	P	1
<i>Regiae</i>	<i>N. alata</i>	Philippines	P	1
<i>Regiae</i>	<i>N. burbidgeae</i>	Borneo	0	1
<i>Regiae</i>	<i>N. chaniana</i>	Borneo; (SH)	P	*, 1
<i>Regiae</i>	<i>N. clipeata</i>	Borneo; (SH)	P	*, 1
<i>Regiae</i>	<i>N. deaniana</i>	Philippines (Palawan)	P	1
<i>Regiae</i>	<i>N. ehippiata</i>	Borneo	P	1
<i>Regiae</i>	<i>N. eymae</i>	Sulawesi; (SH)	P	*, 1
<i>Regiae</i>	<i>N. faizaliana</i>	Borneo	P	1
<i>Regiae</i>	<i>N. fusca</i>	Borneo (Kinabalu)	P+M	1
<i>Regiae</i>	<i>N. graciliflora</i>	Philippines; (SH)	M	*
<i>Regiae</i>	<i>N. klossii</i>	New Guinea; (AW)	0	*
<i>Regiae</i>	<i>N. lowii</i>	Borneo (Mulu)	P	1
<i>Regiae</i>	<i>N. maxima</i>	cult. (SH) & New Guinea (Poso)	P	*, 1
<i>Regiae</i>	<i>N. oblanceolata</i>	Papua; (SH)	P	*, 1
<i>Regiae</i>	<i>N. philippinensis</i>	Philippines (Palawan)	P	1
<i>Regiae</i>	<i>N. spec.</i>	Borneo (E Kalimantan)	P	1
<i>Regiae</i>	<i>N. stenophylla</i>	Borneo (Bario)	P	1
<i>Regiae</i>	<i>N. veitchii</i>	Borneo; (Bario, “highland”); (SH)	0	*, 1
<i>Regiae</i>	<i>N. viridis</i>	Philippines (Dinagat); (SH)	M	*
Artificial Hybrids				
<i>Regiae</i> × <i>Insignes</i>	<i>N. alata</i> × <i>ventricosa</i>	P × P; <i>Nepenthes</i> × <i>ventrata</i> (SH)	P	*
<i>Regiae</i> × <i>Villosae</i>	<i>N. burbidgeae</i> × <i>edwardsiana</i>	0 × M; (AW)	M	*
<i>Insignes</i> × <i>Regiae</i>	<i>N. campanulata</i> × <i>maxima</i>	0 × P; (AW)	P	*
<i>Regiae</i>	<i>N. chaniana</i> × <i>veitchii</i>	P × 0; <i>Nepenthes</i> “ <i>pilosa</i> ” × <i>veitchii</i> (SH)	P	*
<i>Regiae</i>	<i>N. clipeata</i> × <i>eymae</i>	P × P; (AW)	P	*

Table 1. Continued.				
Section ^a	Taxon	Provenance/comment ^b	Quinone(s) ^c	Ref. ^d
<i>Regiae</i>	<i>N. eymae</i> × <i>clipeata</i>	P × P; P. Debbert, Munich ca. 1989 (SH)	P	*
<i>Tentaculatae</i> × <i>Insignes</i>	<i>N. hamata</i> × <i>campanulata</i>	P × 0; (AW)	P	*
<i>Regiae</i> × <i>Insignes</i>	<i>N. lowii</i> × <i>campanulata</i>	P × 0; (AW)	P	*
<i>Regiae</i>	<i>N. lowii</i> × <i>ephippiata</i>	P × P; (AW)	P	*
<i>Regiae</i> × <i>Insignes</i>	<i>N. lowii</i> × <i>merrilliana</i>	P × 0; (AW)	P	*
<i>Regiae</i> × <i>Montanae</i>	<i>N. lowii</i> × <i>spectabilis</i>	P × 0; (AW)	P	*
<i>Regiae</i> × <i>Villosae</i>	<i>N. lowii</i> × <i>truncata</i>	P × 0; (AW)	P	*
<i>Regiae</i> × <i>Pyrophytae</i>	<i>N. maxima</i> × <i>northiana</i>	P × P; <i>Nepenthes</i> × <i>mixta</i> ; (SH)	P	*
<i>Montanae</i> × <i>Villosae</i>	<i>N. talangensis</i> × <i>truncata</i>	P × 0; Weil am Rhein 2007 (SH)	P	*
<i>Villosae</i> × <i>Regiae</i>	<i>N. truncata</i> × <i>ephippiata</i>	0 × P; (AW)	P	*
<i>Regiae</i> × <i>Villosae</i>	<i>N. veitchii</i> × <i>edwardsiana</i>	0 × M; Weil am Rhein 2014 (SH)	M	*
<i>Regiae</i> × <i>Villosae</i>	<i>N. veitchii</i> × <i>edwardsiana</i>	0 × M; Weil am Rhein 2018 (SH)	M	*
<i>Regiae</i>	<i>N. veitchii</i> × <i>lowii</i>	0 × P; (AW) and Weil am Rhein 2012 (SH)	P	*
<i>Regiae</i>	<i>N. veitchii</i> × <i>maxima</i>	0 × P; H. Hennern (SH)	P	*
<i>Insignes</i> × <i>Urceolatae?</i>	<i>N. ventricosa</i> × <i>mapuluensis</i>	P × 0; (AW)	P	*
<i>Insignes</i> × <i>Urceolatae?</i>	<i>N. ventricosa</i> × <i>mapuluensis</i>	P × 0; (AW)	P	*
<i>Insignes</i> × <i>Regiae</i>	<i>N. ventricosa</i> × <i>maxima</i>	P × P; (see reference)	P	5

^aClassification according to Clarke *et al.* 2018

^bPlants investigated in this study: AW = cultivated by Andreas Wistuba; SH: cultivated by Siegfried & Irmgard Hartmeyer

^c**P**: plumbagin, **M**: ramentaceone, **0**: no quinone detected

^d * New/additional data from this study

1 Schlauer *et al.* 2005; all investigated specimens cultivated by Joachim Nerz

2 Rischer *et al.* 2002 A hybrid origin of the studied plants cannot be excluded. Possible partners with **P** and putatively overlapping distribution are *N. mirabilis*, *N. papuana* and *N. maxima* (H. Rischer pers. comm.).

3 Likhitwitayawuid *et al.* 1998

4 Aung *et al.* 2002

5 Shin *et al.* 2007

Discussion

Like in the related family Droseraceae, naphthoquinone patterns are fairly constant within species, and some isomer preference is observed in most sections of *Nepenthes*. **P** is predominant in sections *Nepenthes* (present in 91% of the investigated species, vs. 17% **M**), *Urceolatae* (57% **P**, 0% **M**), *Tentaculatae* (100% **P**, 0% **M**), *Montanae* (68% **P**, 36% **M**), and *Regiae* (74% **P**, 16% **M**), **M** is predominant in section *Insignes* (67% **M**, 22% **P**), and both isomers are present without clear predominance in sections *Villosae* (44% each) and *Pyrophytae* (53% **M**, 38% **P**).

In spite of the anticipated high heterozygosity (frequent introgression, high ploidity) of most *Nepenthes* taxa, only 9 of 96 studied species (9.4%, with no obvious concentration in any section) contain both isomers in the same plant. This proportion is only slightly larger than in Droseraceae (19 of 214 investigated taxa, 8.9%), which may indicate selection against heterozygosity in this trait. Unfortunately, no artificial hybrid between quinone-heterogenous parent species was available in this study. But it is anticipated that such hybrids would contain both isomers like in the related genus *Drosera* (Schlauer & Fleischmann 2016).

All investigated hybrids contained their parents' naphthoquinone isomers, and in particular crosses with naphthoquinone-free species did not yield additional isomers. This strongly suggests that the ability to direct naphthoquinone biosynthesis towards one of the isomers (divergent regioselectivity) is inherited together with the ability to produce naphthoquinones (common polyketide synthesis and subsequent redox/cyclization reactions), and both abilities (that are most probably dependent on separate enzymes, cf. scheme 1) are obviously lost in species that do not contain any isomer.

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MICROSCOPIC FEATURES USEFUL FOR IDENTIFYING *UTRICULARIA* NATIVE TO THE WESTERN USA, WITH AN EMPHASIS ON THE BLADDER QUADRIFID GLANDS

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Introduction

Identifying species of *Utricularia* can be challenging, especially when the plants are not in flower. Even in the western USA, where relatively few species occur, a non-flowering mass of *Utricularia* shoots floating in the water can be difficult.

Of course, flowering *Utricularia* can be identified relatively easily. Out of flower, the patient student can identify species by a number of vegetative characteristics (shoot dimorphism, leaf segment flattening, and leaf setulae being key). However, when flowers are not present, or the collected specimen is incomplete, or the leaves are hopelessly mired in muck (as often the case on herbarium specimens) a confident identification can be challenging.

Fortunately, the bladders can be used to help in identifications. On the interior walls of the bladders one can find peculiar, four-armed glands that are called the quadrifid glands (Figs. 1A-F). Occasionally, one may also encounter two armed (bifid) glands (Fig. 1C). The glands are tiny—even the largest I have observed are less than 120 microns long—about 0.1 mm, so must be observed with a compound microscope at magnifications of 100-1000×. The function of these glands is a matter of contention, but probably is associated with removing fluid from inside the bladder, as well as carnivorous functions (Taylor 1989).

This paper is a primer on how to use the bladder glands to identify species in the western states of the USA, and is a useful supplement the botanical treatment I wrote for the California Flora (Rice 2012). Not only do the quadrifid glands have diagnostic features that are unique to each species, but these features can be used to identify both freshly collected, live specimens and also rehydrated herbarium samples.

Preparing specimens

Preparing *Utricularia* bladders for microscopy is a delicate process. Live or dead, the easiest bladders to work with are the largest you can find. Also, it is best if the bladders are as free of internal detritus or prey as possible. Extremely heavily pigmented bladders can also be difficult to see through—*Utricularia macrorhiza* bladders are sometimes purplish to nearly black, so look for younger bladders that have not yet pigmented. Live or dead, the specimens must usually be prepared under a dissecting microscope.

When I am examining live bladders, I usually quickly look at them under a compound microscope, under a cover slip, with no prior preparation—often the bladder glands can be seen with no further work. Otherwise, the nearly spherical bladder must be reduced to a specimen consisting of

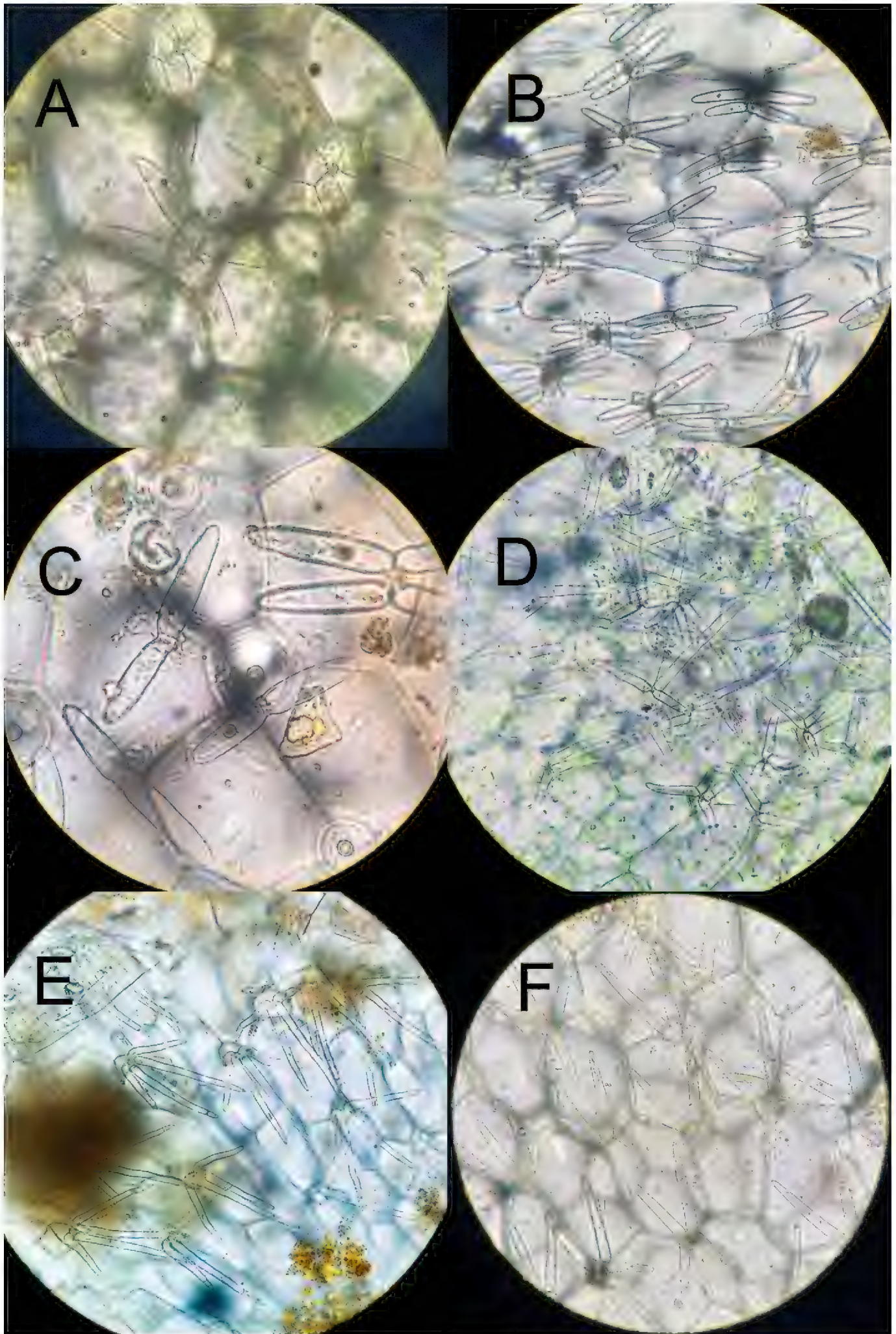


Figure 1: *Utricularia* glands. A: *U. gibba*, 1000x, Nevada County, California; B: *U. intermedia*, 400x, Butte County, California; C: *U. intermedia* (note the bifids), 1000x, Butte County, California; D: *U. macrorhiza*, 400x, Lassen County, California; E: *U. minor*, 400x, Lassen County, California; F: *U. ochroleuca*, 400x, Lassen County, California.

its flattened walls. If this is the case, use one or two needles to remove the trap opening by crushing, cutting, and tearing. Next, split the trap open, splaying the walls into two halves—a bladder fillet.

Despite the violence of this process, the majority of the quadrifid glands will retain their critical characteristics. Sometimes this preparation process is quite destructive, and only small portions of the trap walls survive the process...more than one bladder may have to be dissected. Once the specimens are prepared, they are placed under a coverslip.

When I am examining dried herbarium specimens, only a few bladders must be removed for examination. (Destructively sampling an herbarium specimen—even just removing a few bladders—requires permission from the owner of the specimen.) Sometimes loose specimen fragments are included on an herbarium sheet in an attached envelope, but even if not, usually a few loose bladders can be found here or there on the herbarium sheet.

Once the dried, flattened bladder has been removed, I look at it under a compound microscope, in case quadrifids can be seen with no further dissection. But if the bladder must be dissected, it can be using the method described as for live specimens. A different method, that I learned from Jan Schlauer, is to sandwich the bladder between two pieces of transparent tape. Then, by carefully separating the tape, each piece of tape carries away one separate bladder wall, ready for microscopy!

I examine the bladder specimen while it is still dry, then I moisten the specimen and examine it again. Almost invariably, in either dry or wet conditions, the quadrifid glands can be detected. While somewhat shriveled, their key characters can be observed. The results of my observations are always noted on the herbarium specimen as an annotation.

Key characters

Structurally, quadrifid glands usually have two pairs of oppositely directed gland arms. One pair is often consistently shorter than the other pair—hence the terms long arms and short arms. The two arms in a pair may be essentially parallel (diverging from each other by 0-30°) or by as much as 180°. In some cases, the arms may diverge by even more than 180°, so that both arm pairs are pointing in the same general direction.

While quadrifids may be useful in making identifications in combination with other observed features of a specimen, in the western USA, quadrifids by themselves can be used quite effectively to key a plant to species in nearly all cases. What follows is a key based purely upon quadrifid gland characters. When using this key, it is important to look at several examples of glands in a trap, and use the average or typical gland characteristics...it is always possible that a single gland may be distorted by the specimen preparation process.

- 1A: The angle between the short arms is 90° or more.....2
- 2A: The angle between the short arms is 90-180°.....3
- 3A: Short arms are less than ½ the length of the long arms; long arms are up to 120 microns long; long arms as much as 13× longer than wide.....*U. macrorhiza*
- 3B: Short arms are ½ to 1× the length of the long arms; the long arms are less than 50 microns long; long arms are 6-10× longer than wide *U. ochroleuca* (1)
- 2B: The angle between the short arms is 270-300° *U. minor*
- 1B: The angle between the short arms is 0-80° 4
- 4A: Long arms and short arms are in parallel pairs, diverging by 0-30°*U. intermedia*
- 4B: Long arms and short arms diverge by more than 30° 5

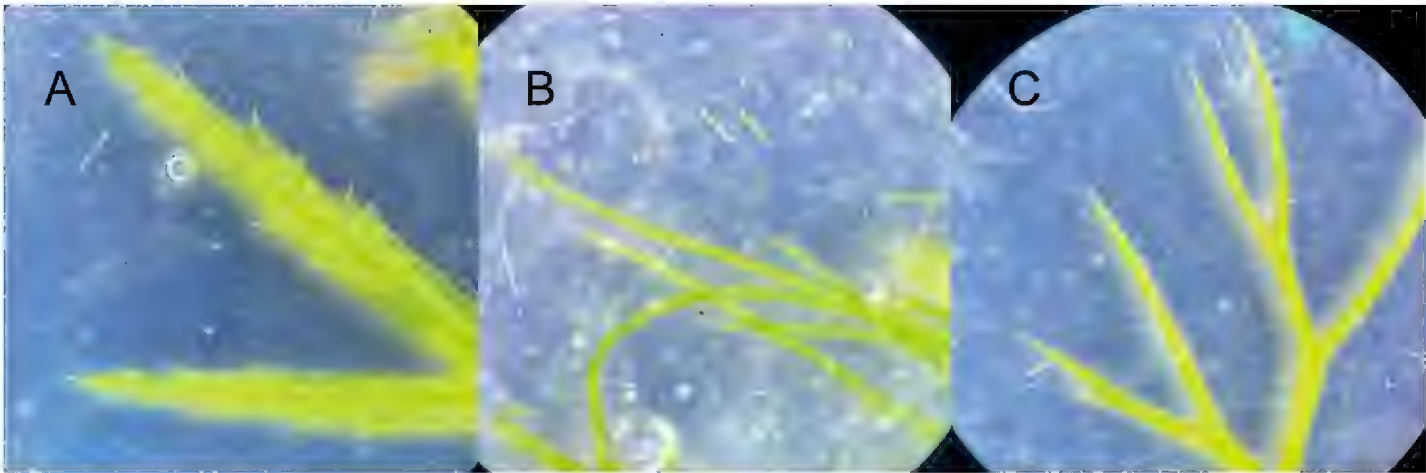


Figure 2: A: *Utricularia ochroleuca*; B: *Utricularia macrorhiza*; and C: *Utricularia minor* leaf segment tips at 45x. All specimens from Lassen County, California.

- 5A: All arms are approximately the same length; all arms are 1.5-6× longer than wide; both arm pairs diverge by approximately the same angle..... *U. gibba*
- 5B: A long and short arm pair dimorphism is clearly present; the long arms are 6-10× longer than wide; the long arm pair diverges by 20-45° while the short arm pair diverges by 40-160°..... *U. ochroleuca* (2)

This key is fairly straightforward, except for challenges with *Utricularia ochroleuca*, which has somewhat unremarkable glands. As such, couplets 3 and 5 above can be difficult to apply. Fortunately, even inadequate collections can easily resolve *Utricularia ochroleuca* from *U. macrorhiza*, and *Utricularia ochroleuca* from *U. minor* on other characters such as leaf morphology, shoot dimorphism, and bladder size. In particular, the leaves of *U. ochroleuca*, *U. macrorhiza*, and *U. minor* are distinctive. On the ultimate leaf segments (which are flattened), *Utricularia ochroleuca* bears numerous tiny spinelike hairs (setae) on the leaf margins, and the hairs are borne at the tips of tiny lateral teeth (Fig. 2A). In contrast, the ultimate leaf segments of *U. macrorhiza* are hairlike (not flattened), and the setae are larger but are not borne on tiny marginal teeth (Fig. 2B). Finally, the leaf segments of *U. minor* are at least slightly flattened, and almost never bear setae on the leaf margins (Fig. 2C) —although they occasionally are borne on the leaf segment tips.

Species comments

Utricularia gibba (Fig. 1A)

In my home state of California, this is a confounding species. It occurs at high elevations in the central Sierra Nevada (Tuolumne to Fresno Counties), and also at scattered coastal sites—these appear to be native populations. Meanwhile, additional populations appear sporadically in the Central Valley, especially in rice farming operations in Butte County. It is unclear if these populations are native or introduced, and how they are moving from site to site—either by agricultural equipment or waterfowl.

The quadrifid arms are small (10-40 microns long), with two pairs of more or less similar arms, weakly diverging. The arms are long-ovoid with acute tips. The bladders on this species are never large.

Utricularia intermedia (Figs. 1B, 1C)

This species produces its bladders on fragile, white or transparent shoots that affix the plant to the muck, and which require care to remove. Accordingly, hastily collected specimens often lack

bladders-bearing shoots. However, if they are present, the bladders—which are very large—are easy to work with.

The quadrifids arms are fairly small (approximately 50 microns long), with two pairs of nearly parallel arms. You will frequently see weakly diverging arms, but you will also see pairs of arms appressed tightly against each other—a feature you will never see in other species. The arms are uniformly wide over most of their lengths, and are usually blunt tipped.

Utricularia macrorhiza (Fig. 1D)

Very often, botanists not experienced with the genus trying to identify small plants will call it “*Utricularia minor*” based on stature alone, but under less than ideal conditions *Utricularia macrorhiza* can be quite small. Even dwarfed or stressed, the quadrifid gland morphology is reliable.

This species is usually very easy to work with. The dimorphism between arm pairs—long vs. short, and weakly diverging vs. strongly diverging—is easily visible. The angle between the short arms can be variable, as is seen in Figure 1D. The long arms are large (up to 120 microns long). The arms weakly tapering, and blunt tipped.

Utricularia minor (Fig. 1E)

Despite typically bearing small bladders, the quadrifids of this species are usually very visible. The quadrifid arms are large (about 100 microns), which is remarkable considering the bladders may be less than 1 mm (1000 microns) in size! The arms are very gently tapering, and acutely tipped.

Utricularia ochroleuca (Fig. 1F)

This species is generally considered to be of hybrid origin, and it is debatable whether it should be considered a true species, or simply a sporadic hybrid that is at best an intriguing curiosity. The plant is very similar to *U. intermedia*, but is often a little less robust. The quadrifid arms are moderately small (50 microns), are weakly tapering, with acute tips.

Note that Figure 3 is a rehydrated herbarium specimen, and despite having been dried, the critical quadrifid gland characteristics have been retained.

Work by Thor (1988) suggests that *Utricularia ochroleuca*, in the broad sense, is actually a mix of two species—*Utricularia ochroleuca* and *U. stygia*. Among the differences in these two species are subtleties in quadrifid structure. Schlosser (2003) used these differences to conclude that at least some of the populations of *Utricularia ochroleuca* in California should be considered *U. stygia*. Schlosser summarized the quadrifid glands of the two putative taxa as follows:

Utricularia ochroleuca: long arms separated by 18-52°, short arms separated by 146-196°. *Utricularia stygia*: long arms separated by 26-56°, short arms separated by 52-96°.

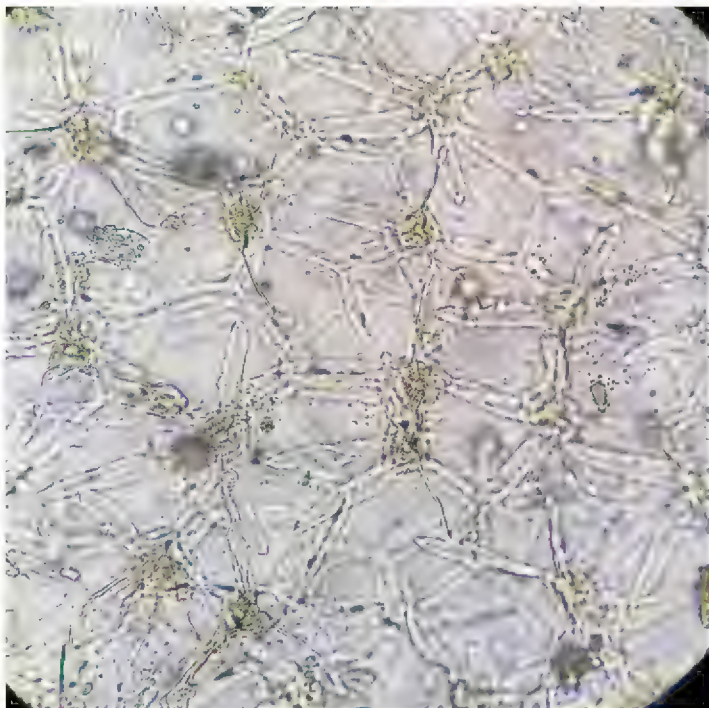


Figure 3: *Utricularia ochroleuca*, 400×. Rehydrated specimen, Teton County, Wyoming.

Generally speaking, *Utricularia stygia* quadrifids would have a structure more similar to *U. gibba*, while *U. ochroleuca* (more narrowly described) would look more similar to *U. macrorhiza*. Whether one considers these significant on a species level, or even if *U. ochroleuca* (broadly described) itself should be treated as a species, is a matter for discussions and fistfights.

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VARIATION IN *TRIANTHA OCCIDENTALIS* (S.WATSON) R.R.GATES CHARACTERISTICS IN CALIFORNIA

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In case you haven't noticed, recent research (Lin *et al.* 2021) indicates that the showy monocot *Triantha occidentalis* may be carnivorous. *Triantha occidentalis* contains three subspecies—*T. o.* subsp. *occidentalis*, *T. o.* subsp. *brevistyla*, and *T. o.* subsp. *montana* (Rice 2021). In their paper, Lin *et al.* focused their *Triantha* work on Canadian specimens of *Triantha occidentalis* subsp. *brevistyla* (S. Graham, pers. comm. 2021). However, they included images of *Triantha occidentalis* subsp. *occidentalis* in their discussions, indicating that they are concluding carnivory occurs in all the subspecies of *Triantha occidentalis*.

In a previous paper (Rice 2022) I provided observations and imagery that I made on a trip to Washington state. They appear to support evidence of carnivory in *Triantha occidentalis* subsp. *brevistyla*—the plants occupied the same habitats as carnivorous plants, had impressively sticky red glands on the flowering stems, and were capturing insects (Fig. 1A).

In my home state of California, we have the southern subspecies, *Triantha occidentalis* subsp. *occidentalis*. Broadly speaking, *Triantha* in California occurs in two large range segments. One segment is in the northwest corner of the state. The other segment is a long strip following the Sierra Nevada, from Shasta County southeast to Tulare County. There are also, strangely, a few disjunct locations on the coast in central California. In this paper I present a few observations of this plant that I made at four populations at three separate locations.

The first site was in Marin County, at Point Reyes National Seashore, at one of the disjunct coastal sites. The plants occur in a heavily grazed area, where freshwater seeps through the

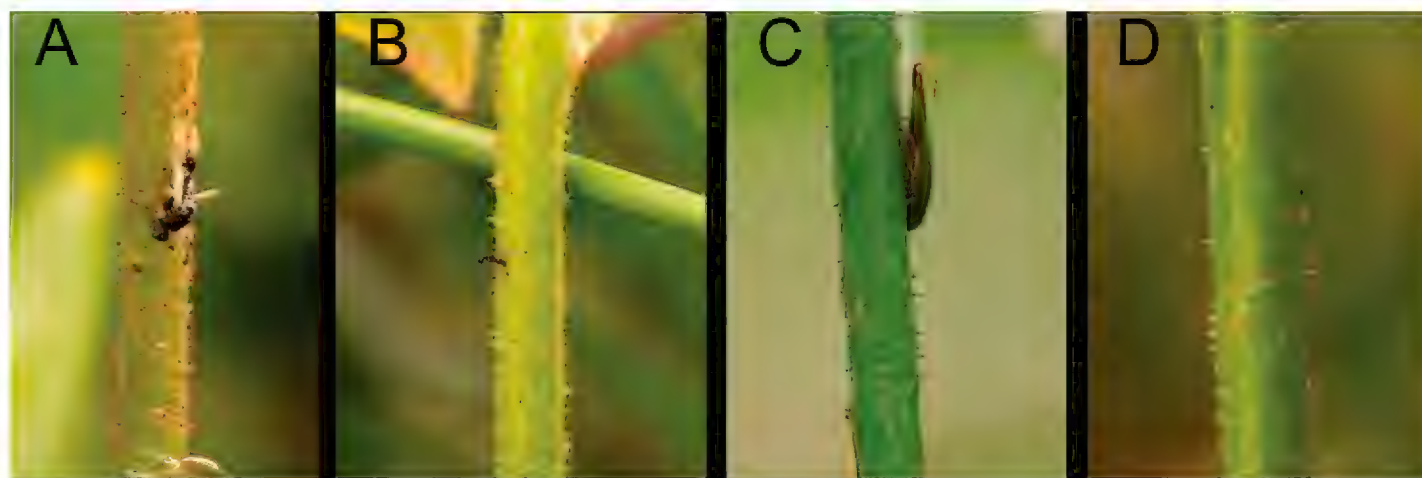


Figure 1: Inflorescence glands just below the lowest flower or fruit. A) *Triantha occidentalis* subsp. *brevistyla*, Whatcom County, Washington; B) *Triantha occidentalis* subsp. *occidentalis*, Marin County, California; C) *Triantha occidentalis* subsp. *occidentalis*, western slope Nevada County, California; D) *Triantha occidentalis* subsp. *occidentalis*, eastern slope Nevada County, California.

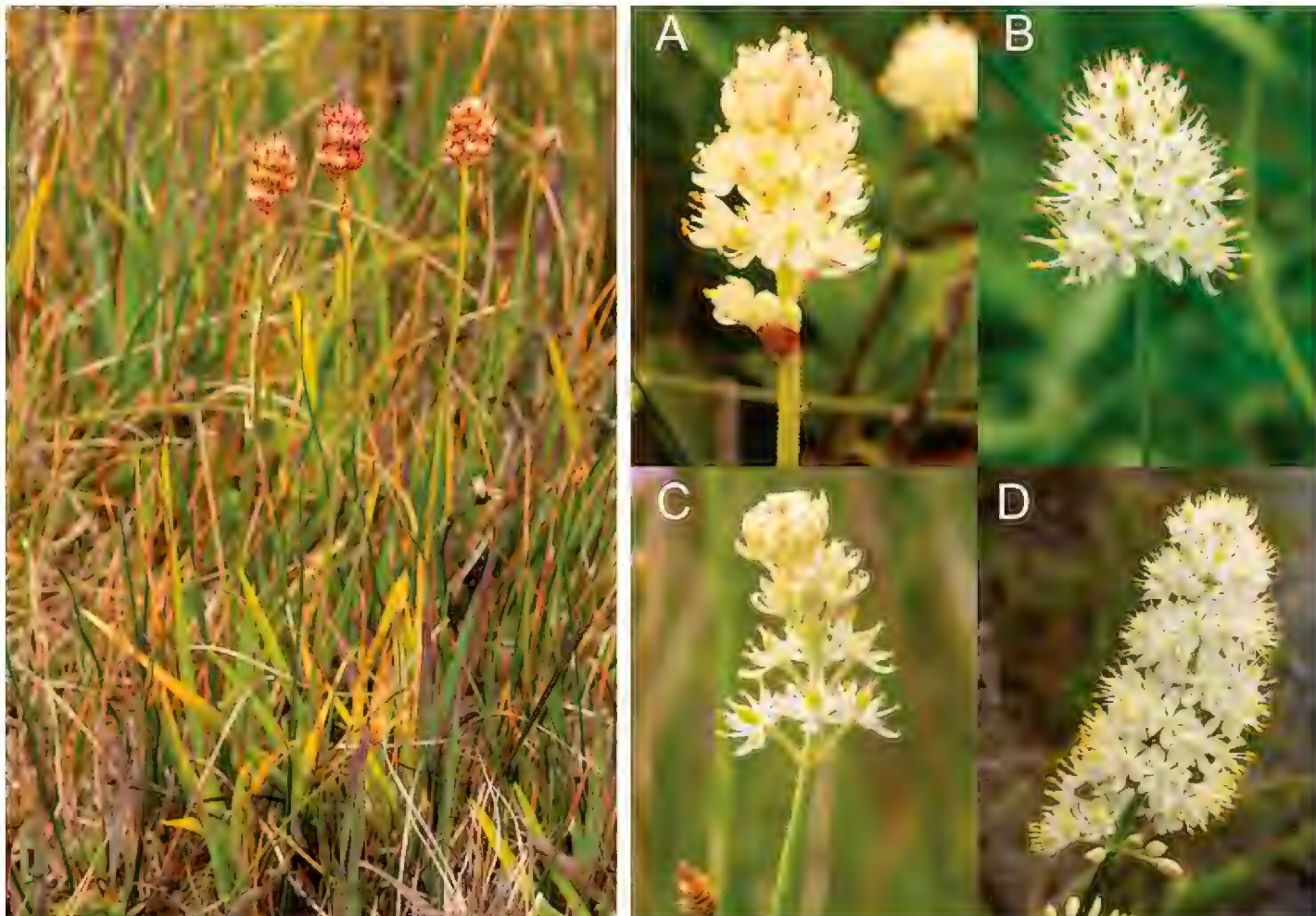


Figure 2: (left): Compact fruiting *Triantha occidentalis* subsp. *occidentalis* in Marin County, California.

Figure 3: (right): Inflorescence heads of *Triantha occidentalis* subsp. *occidentalis* in California. A) Marin County; B) western slope Nevada County; C) eastern slope Nevada County; D) Del Norte County.

muck into an estuary, essentially at sea level. No carnivorous plants occur in the area, but other plants that often occur with carnivorous plants in California (such as the orchid *Spiranthes romanzoffiana*) were present. In addition to the population I visited, at least one other smaller colony exists in the area (A. Yin, pers. comm. 2022). Plants were compact and relatively short (typically flowering stems were less than 30 cm tall) and many showed indications of browsing—cattle and deer are common in the area. The inflorescences were also very compact and capitate (headlike). Point Reyes is a very windy place, and much of the vegetation there responds by adopting a compact habit—this might explain the short stature of the *Triantha* there (Fig. 2).

Generally speaking, the plants in Marin County (Fig. 1B) exhibited the same glandular structure as the specimens of *Triantha occidentalis* subsp. *brevistyla* that I observed in Washington. While the glands were as densely packed as they were in the Washington plants, they did not seem to be quite as brightly pigmented. Minute, trapped insects were frequently observed.

The second site I visited was in a wet seepage in Nevada County, at an elevation of about 1800 m, on the western slopes of the Sierra Nevada. This site is notable for its population of *Darlingtonia californica* f. *viridiflora*, as well as *Drosera rotundifolia*. The *Triantha* plants here were nearly twice as tall as the plants in Marin County, and the inflorescences were commensurately larger and more delicate (Fig. 4) although still very capitate (Fig. 3B). The glands on the inflorescences were more scattered, and less brightly pigmented (Fig. 1C). While some of the flowering stems had



Figure 4: Tall flowering *Triantha occidentalis* subsp. *occidentalis* in Nevada County, California, on the western slopes of the Sierra Nevada. The large dried infructescences belong to *Bistorta bistortoides*.

bits of detritus stuck to them (seeds, dirt, etc.), none of them were observed to have what could be clearly identified as captured insects.

The third location I visited was also in Nevada County, at an elevation of 2000 m, on the eastern slopes of the Sierra Nevada. There were two separate *Triantha* populations here. One was in an area subject to seasonal dry periods—while it was moist enough for some wetland plants such as the orchid *Platanthera dilatata* var. *leucostachys*, or *Erythranthe primuloides*, it had no carnivorous plants. It was the densest population of *Triantha* I have yet observed (Fig. 5), and the plants were large and diffuse, similar to the plants at the previously described Nevada County site. Interestingly, the glands on these plants were more sparsely scattered than I observed on any other plants. Furthermore, they were essentially lacking in red pigmentation while in flower, although they developed some red pigmentation when in fruit (Fig. 1D). The second population of plants occurred in a nearby perennial seepage that also housed *Drosera rotundifolia*, *D. anglica*, *D. × obovata*, and *Utricularia minor*. These plants were slightly shorter and somewhat more glandular. The plants at both sites had comparatively more elongate inflorescences (Fig. 3C). Plants from the northwestern portion of California have even larger, more elongated inflorescences, as can be seen in an image I took on a previous trip to Del Norte County (Fig. 3D).

The carnivorous character of *Triantha occidentalis* is certainly quite variable, and should be studied more. It would be interesting to determine if all the varying forms of *Triantha occidentalis* subsp. *occidentalis* are as carnivorous as *Triantha occidentalis* subsp. *brevistyla*. It would also be interesting to carefully examine the plants in the northwestern part of the state, especially since some of the plants in that part of the state are particularly large.



Figure 5: A dense population of *Triantha occidentalis* subsp. *occidentalis* in Nevada County, California, on the eastern slopes of the Sierra Nevada.

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HUNTING THE KING – *DROSERA REGIA* TRIP REPORT

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Drosera regia is one of the most iconic *Drosera* species, popular for its impressive stature and rarity. While most sundews are relatively small, *D. regia* clocks in with impressive 50 cm+ sharply pointed leaves that point skywards, making large specimens a sight to behold. It is also noted for its ability to capture copious volumes of insects and is the most basal *Drosera* species, the closest sundew relative to Venus flytraps (*Dionaea muscipula*) source. Despite its fame and reputation, little work has been done on the species in the wild. As it turns out, *D. regia* is one of the rarest South African sundews. In June 2022 I trekked out to check on these elusive giants and answer some burning questions about them.

While sometimes seen in nurseries, *Drosera regia* is exceptionally hard to find in South Africa’s wilds. It is endemic to the Cape Fynbos, a floristic kingdom found only in Mediterranean regions of the country, famous for the insane diversity of plants, sundews included (Fig. 1). To add to this local limitation, the species is only found along Bain’s Kloof Pass (also Bainskloof Pass), a mountainous pass between the towns of Wellington and Slanghoek built by road engineer Andrew Bain, which opened in 1853. No other localities are known, and for this reason it is listed as “rare” in the SANBI redlist. Site details are closely guarded. Currently, three locations are known: a lower location, the “upper waterfall” site, and another site higher in the mountains. The first two sites are both



Figure 1: My brother helping me scour the Fynbos for *Drosera regia*.

cultivated, the latter was discovered recently by a hobby botanist! *Drosera regia* habitat is typically described as peaty gravel soils with water running through them, and typically dominated by various plants in the family Restionaceae (one of the major fynbos groups) which makes the sundews quite hard to find, as both are tall plants with thin leaves! Most records I could find occur early in the year during summertime when the plant flowers. It would be hard to miss the large pink blooms erected above the surrounding vegetation. It is also known to go dormant, as seen in cultivation, but nobody knows exactly when. There is a gap in records after March in late Summer (after flowering) and before September, about springtime (emerging growth). Between this is a chasm of mystery as to what happens with the plant.

It doesn't help that the pass was mostly closed for over two years for roadwork and maintenance. I was due to join Alex Dietrick to see them October 2021, but it rained so much before the trip that the Tweede Tol campsite closed due to damage! I briefly visited the pass to see a new *D. rubrifolia* population in January 2022, but time was too tight to see *D. regia* as well.

To figure out what *D. regia* is up to there was no better way than to march into the mountains, camera in hand. It was a bright June day, and surprisingly warm for what is supposed to be winter. It took some river crossing and hiking along rocky Cape trails (they are rarely even or flat) to reach the lower site, a lovely wetland patch along a gentle slope. It was peaty and wet with many open patches, absolutely perfect for *Drosera*. Despite that, I could not find any! Even with a few sets of extra eyes and two different coordinates from reputable sources, not a single regia was found! I suspect the mountain nights were significantly cooler than the days, so the plants were likely dormant. A bummer after the hike, but no data is still data! I suspect they may go dormant soon after setting their seed, similar to winter growing sundews like *Drosera cistiflora*. I did not pursue the higher populations, as they were likely dormant too. They were seen at this site last year, so I am sure they are still around!

Another concern with this rare species is encroachment of invasive plants, or anthropogenic interference in the fire regime, something that fynbos as a biome (and many carnivorous plants) rely on for proper succession and function. Without fire, it is possible that surrounding vegetation could grow too densely and choke out the light-loving sundews (it is in the name!), a phenomenon I have seen occur on Table Mountain that has not seen burns in decades. Thankfully for our sticky buddies in Bainskloof, a natural fire ripped through its habitat in late 2017, clearing much of the overgrowth and allowing the plants to thrive. I could see this in the habitat, with many open patches allowing sundews to proliferate and grow readily. Thanks to hacking efforts I didn't see a single pine anywhere near the habitat, although they are still present in the general area.

Despite the disappointment in not finding *D. regia*, I did still find some truly wonderful sundews! *Drosera admirabilis* is a small rosetted species, also a Cape endemic, known for its beautiful round leaves and intense colours (Figs. 2 & 3). It was introduced to cultivation from Bainskloof, initially distributed as "species floating" as it is often observed almost submerged in shallow pans of water, or "floating"! It was described by Debbert in 1987, after being in cultivation for some time before that. If you see a sundew sold as "sp floating" or "admirabilis holotype" it is *Drosera admirabilis* from Bainskloof! It is exciting to see a species in its location of discovery, or to find the source of a legendary plant like seeing the patch of *D. capensis* that all Gifberg location plants originate from! These plants were in almost every wet, peaty patch that you could see along the seepage, often forming large clumps in exposed areas. I even saw some growing out of fissures in small rocks. Several plants were lying exposed on substrates, potentially washed out or having their habitat eroded by rain sometime before. They had substantial root systems, some up to 15 cm long. I can't say I have seen them growing semi-submerged anywhere but Bainskloof, usually on moss banks or small seepages on paths. It, like *D. capensis*, is one I only see in heavily waterlogged substrates.



Figure 2: Hendré with some *Drosera admirabilis* in situ.



Figure 3: Typical habitat of *Drosera admirabilis* in Bainskloof.

While I did not find the king of sundews, this trip was both gorgeous in views and valuable for the information on dormancy and habitat of the species. It will take many repeat visits and possibly camera monitoring to really understand the niche and habits of *D. regia*. Nothing good ever comes easy as they say. Regardless, it is comforting to know that the species habitat is still secure and that I have an excuse to hike this amazing area many more times in future! Looking forward to finally seeing the elusive behemoth in the wild. I hope to return in September to look for new locations of a novel species in the *D. cistiflora* complex, I will make some pitstops to the king and see if they have woken up by then. So much discovery to do!

NEW CULTIVARS

Keywords: cultivar, *Dionaea* ‘EEC Skyscraper’, *Dionaea* ‘F.S. Loony Clam’, *Sarracenia* ‘Purple Devil’, *Sarracenia* ‘Vanilla Swirl’, *Pinguicula* ‘PE Formosa’.
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Abstract: Five new carnivorous plant cultivars are named and described: *Dionaea* ‘EEC Skyscraper’, *Dionaea* ‘F.S. Loony Clam’, *Sarracenia* ‘Purple Devil’, *Sarracenia* ‘Vanilla Swirl’, *Pinguicula* ‘PE Formosa’.

Sarracenia ‘Purple Devil’

Submitted: 20 July 2022

In 2017, a cross between *Sarracenia oreophila* (female plant) and *Sarracenia purpurea* subsp. *purpurea* (dark form) was made. Unfortunately, only one plant survived.

Sarracenia ‘Purple Devil’ (Fig. 1) has slightly veined pitchers in spring like *Sarracenia oreophila* but with a larger lid. In early summer, the purple coloring appears on the upper part of the pitcher tube and hides the veinlets. The pitchers are medium sized (50 cm tall), as is the peduncle. The petals are yellow-orange in colour. Note that the plant produces many phyllodes at its base like *Sarracenia oreophila*.

Sarracenia ‘Purple Devil’ must only be propagated vegetatively to preserve its unique characteristics.

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Figure 1: *Sarracenia* ‘Purple Devil’ spring pitchers, fall pitchers, and flower (left to right).

Submitted: 31 August 2022

Dionaea ‘EEC Skyscraper’ is the product of hand pollination in the summer of 2018 by Evan and Emmy Wang (Ev & Em Carnivorium, Palo Alto, California, USA). After pollination, isolation of flowers was performed. The seed was the product of *D.* ‘Phalanx’ × an unnamed *Dionaea* selected for its long cilia. Numerous seeds of this cross were grown by Evan and Emmy Wang. Of the over 60 seeds, only one developed the unique phenotype of *D.* ‘EEC Skyscraper’.

Dionaea ‘EEC Skyscraper’ (Fig. 2) is characterized by extremely long petioles, reminiscent of *D.* ‘WIP Slim Snapper’, but the cilia are much longer and the trap morphology is box-like. Traps and petioles are green with red interior when grown in direct sunlight. Traps are symmetric and fully functional. Trap efficiency is very high as we have observed nearly all traps on our plants with digested prey. Trap speed appears to be slowed, possibly by the extremely long cilia, but the cilia length seems to make up for the loss of speed as we have seen high rates of trapped prey. This cultivar is extremely vigorous and divides readily. Trap length on adult plants range up to 3.5 cm with a box-like morphology. Elongated cilia can measure up to 2 cm and the cilia length to trap length ratio is typically 2:3. Petioles are thin and delicate, measuring up to 13 cm in length. Petiole growth is often nearly vertically upright in the spring and eventually more lateral spread as the growing season continues into the summer months.

The name ‘Skyscraper’ refers to the vertical growth of this cultivar in the spring with extremely long petioles; similar to a very tall building. The ‘Scraper’ portion of the name also references the very long cilia which seemingly scrape the air as it closes so often on unsuspecting prey.

‘EEC’ is an acronym for Ev & Em Carnivorium where hand pollination, seed germination, and growing were performed. *Dionaea* ‘EEC Skyscraper’ can only be propagated vegetatively by rhizome or leaf/floral cuttings to preserve the unique characteristics of this cultivar. *Dionaea*. ‘EEC Skyscraper’ is currently only grown in Palo Alto, California, USA.

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Figure 2: *Dionaea* ‘EEC Skyscraper’.

Submitted: 20 July 2022

Dionaea ‘F.S. Loony Clam’ (Fig. 3) arose from the propagation of *Dionaea* ‘F.S. Anemone’ by Engineer Juan Camilo Páez Jiménez from the Wild Flora laboratory. We noted it was strikingly different from *Dionaea* ‘F.S. Anemone’.

The plant produces between 4 to 5 upright leaves with an average size ranging between 3 and 4 cm in adulthood. In semi-shade it has a lime green coloration and in direct sun an emerald green coloration. The base of the leaf is elongated with a very marked curvature and a noticeable fold in the middle. As the leaf develops, it begins to thin down toward the trap. It has no petiole, having a direct connection between the base leaf and the trap similar to *Dionaea* ‘Crocodile’. The traps possess an interesting deformity like a clam. At an early age its lobes have square rounded edges without undulations; in adulthood they become more rectangular with striking folds like a giant sea clam shell. Traps do not exceed 1 to 1.5 cm in adulthood. The traps have no teeth, not even a slight vestige. The traps have no trigger hairs.

Dionaea ‘F.S. Loony Clam’ traps have a shape that vaguely resembles those of *D.* ‘Wacky Traps’. The lack of teeth of *D.* ‘F.S. Loony Clam’ somewhat resembles *D.* ‘Adentate’, but has a different shape, size, and coloration. Other cultivars such as *D.* ‘Werewolf’, *D.* ‘Coquillage’, *D.* ‘Cudo’, or *D.* ‘Fondue’ are characteristically differentiated by their type of trap. Each new trap of *D.* ‘F.S. Loony Clam’ can be different from previous ones, making this cultivar distinctive.

As soon as we saw the growth of *Dionaea* ‘F.S. Loony Clam’ from the first 4 months to 3 years, we realized its resemblance to a sea clam, in addition we want to highlight the symbolism with crazy and wild nature in our creations.

To maintain the desired cultivar traits, propagation of *Dionaea* ‘F.S. Loony Clam’ must be done asexually.

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Figure 3: *Dionaea* ‘F.S. Loony Clam’.

Pinguicula ‘PE Formosa’

Submitted: 10 November 2021

Pinguicula ‘PE Formosa’ (Fig. 4) was selected from a batch of seedlings germinated in 2019 in Taiwan. The parentage is undetermined, but it’s most likely a hybrid from *Pinguicula ehlersiae*. This plant stood out from others with its distinct appearance and vigorous growth. The mature plant is about 5 cm wide. The coloration is striking. The rosette is deep red and slightly green near the center bud. Layers and layers of leaves with distinct curvy edges give the plant a unique stereoscopic appearance. Unlike other hybrids I’ve made, one thing that impressed me the most is the plant doesn’t require strong lights to reach such a contrast in coloration, which makes it a perfect plant for both indoor and outdoor growers.

The name “Formosa”, meaning beautiful, was first used in the 16th century during the Age of Discovery by the Portuguese as an adjective, describing Taiwan as a beautiful island at first sight. The idea inspired me as the fabulous plant appearance and the story of the discovery match the description perfectly. “PE” is the initials for my nursery Premium Exotics.

Pinguicula ‘PE Formosa’ must be reproduced vegetatively by leaf pullings to preserve the unique characteristics of the cultivar.

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Figure 4: *Pinguicula* ‘PE Formosa’ plant and flower.

Submitted: 22 September 2022

Sarracenia ‘Vanilla Swirl’ was grown from seed in early 2016 and was selected in the summer of 2019. *Sarracenia* ‘Vanilla Swirl’ (Fig. 5) produces a strong robust pitcher about 60 cm tall. The upper part of pitcher is dominated by white coloration akin to many “Alba” clones. Veins run through the white lid and congregate in a bright pink throat patch, which contrasts against the white peristome, and continue down the pitcher. The peristome is rounded and over pronounced giving the pitchers a circular rounded shape. As the pitchers age, the veins turn the same shade of red as found in the throat and begins to bleed out into the surrounding white pitchers giving a light pink coloration. The flower is pink.

Sarracenia ‘Vanilla Swirl’ is distinctly different from other cultivars, for example, *S.* ‘Iamsatyricon’ has a purple edge to the lid and the peristome turns red, while *S.* ‘Vanilla Swirl’ lacks a purple edge, and the peristome remains white. *Sarracenia* ‘Vanilla Swirl’ is also distinctly different from *S.* ‘Legacy’ by lacking the distinctive edge to the lid, a much lighter throat spot, and a different flower color. *Sarracenia* ‘Lunchbox’ has a much heavier veining segmenting its white lid with a pink peristome, while *S.* ‘Vanilla Swirl’ has less veining in the lid and lacks the pink peristome. *Sarracenia* ‘Leah Wilkinson’ has the same over-pronounced peristome as *S.* ‘Vanilla Swirl’, although in *S.* ‘Leah Wilkinson’ the peristome tends to undulate in the center, while *S.* ‘Vanilla Swirl’ does not. *Sarracenia* ‘Leah Wilkinson’ tends to have a yellow under tone coloration and a clear pink edge to the lid, while *S.* ‘Vanilla Swirl’ lacks the pink edge and has no yellow coloration.

The name of this plant was given because it reminds me of the vanilla ice cream that has had some red sauce added and been mixed together. The analogy to the plant is that the pink coloration begins to bleed from the throat into the white as the pitchers get older and there is no consistency to the pattern, rather, a random mix.

Sarracenia ‘Vanilla Swirl’ must be vegetatively reproduced to retain its unique traits.

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Figure 5: *Sarracenia* ‘Vanilla Swirl’ pitcher from front (left) and top (right). Flower in upper right.

